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SYNTHESIS OF BASIC LIFE HISTORIES OF TAMPA BAY SPECIES

FINAL REPORT

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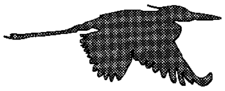

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FOREWORD

This report, *Synthesis of Basic Life Histories of Tampa Bay Species,* was prepared by Versar, Inc under the direction of Dick Eckenrod and Holly Greening of the Tampa Bay National Estuary Program. The work was performed under contract to Versar, Inc. This is Technical Publication #10-92 of the Tampa Bay National Estuary Program.

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EXECUTIVE SUMMARY

OVERVIEW

One of the goals of the Tampa Bay National Estuary Program is to acquire information necessary for maintaining or improving the condition of the estuary's natural resources. The purpose of this project was to characterize habitat types critical to the survival of endemic fish and wildlife species. This information will be used to guide the development of effective strategies for habitat restoration and protection and to identify information necessary for future management decisions.

Determinations of which species were important to the estuary were based on ecological importance, significance to commercial or recreational fisheries, aesthetic value and designations of endangered, threatened, or protected status. Data collected for each selected species were synthesized into individual species summaries, each containing information on life history, ecological role, contaminants and habitat requirements. Both water quality and structural habitats were considered. Water quality habitats included' information on factors such as salinity, water temperature, dissolved oxygen and pH. Structural habitats included information on factors such as bottom substrate, water currents and submerged aquatic vegetation.

An extensive literature search was conducted to identify information necessary for species summaries. During this data acquisition stage, it became apparent that most of the research conducted within the Tampa Bay estuary has been directed at commercially and recreationally important species such as snook, red drum, spotted seatrout, striped mullet and blue crabs. Very little information was available to document environmental requirements of species which can be considered of little economic importance, but which are probably critical to the structure of the ecosystem. These include species such as silver perch, clown goby, striped killifish, grass shrimp and benthic organisms. Some species, including tarpon, bay anchovy, spot, American oyster and hard clam had adequate amounts of information available to identify environmental requirements, although very little was available specific to Tampa Bay.

Two general life history patterns emerged from the species summaries. Some species, including spotted seatrout, clown goby, striped killifish, manatee, hard clam and American oyster remain in the estuary their entire life cycle. Others, such as blue crabs, striped mullet, tarpon, spot, and pink shrimp spawn just outside the estuary or some distance offshore. Currents or tides transport eggs, larvae or juveniles of these species back into the estuary. In either situation, the estuary serves as a nursery area for larval and juvenile stages of these species, and as a forage area for subadults and adults. Degradation of nursery habitats or forage areas within the estuary could seriously affect the populations of many of these species.

Species chosen for this project were representative of many trophic levels within the Tampa Bay estuary. In general, an adequate amount of information existed to characterize feeding ecology of the selected species. However, in many instances, this information was not specific to the Tampa Bay estuary. Ecological studies being conducted by the Florida Marine Research Institute should provide a more thorough examination of the feeding ecology of many of the selected finfish species.

With the exception of American oyster, very little information was available to assess concentrations or effects of contaminants in the estuary. Studies recently initiated by the Florida Marine Research Institute are directed at identifying toxicants in fish tissues and should provide some baseline data in which to direct further research.

Distribution and habitat utilization within the estuary was dependent on species and stage of life cycle. Some species, such as the bay anchovy, had a ubiquitous distribution throughout the estuary, and didn't appear to be influenced by any particular water quality or structural habitat. Many species showed a habitat preference for shallow areas characterized. by submerged aquatic vegetation. Spotted seatrout, silver perch, clown goby, pink shrimp, grass shrimp and the manatee were common residents of these habitats. Backwater areas (including mangrove and marsh areas) and tidal rivers provided critical habitats for many of the selected species. Red drum, snook, striped mullet and spot preferred these habitats, especially during their larval and juvenile stages. More information is necessary to accurately describe the distribution of the benthic organism, *Diopatra cuprea.*

Water quality requirements of many species was variable, depending on life stage and seasonality. Most estuarine species are highly tolerant of fluctuations in water temperature; however, cold water mortalities of snook and tarpon have been observed in Tampa Bay.

Many species have a high degree of tolerance to salinity fluctuations although most exhibited preferences to different salinity regimes. Hogchoker prefer the oligohaline and freshwater portions of tidal rivers. Many of the species were highly tolerant of a wide range of salinities. These included red drum, snook, spot, tarpon, blue crabs, and American oyster. Species such as spotted seatrout, silver perch, lined sole, and striped killifish preferred higher salinities in the meso and polyhaline areas of the estuary. In many estuarine finfish species, tolerance to low salinity waters was greater at juvenile stages and decreased with growth.

Varying amounts of information existed for the remaining environmental requirements, including dissolved oxygen, pH, water current, turbidity or depth preferences. Data suggested that species such as spot, striped killifish, American oyster and hard clams are somewhat tolerant of low dissolved oxygen conditions, depending on the duration of exposure. Juvenile tarpon are able to survive in areas of severe hypoxia. In general, it appeared that preference for the remaining environmental requirements was dependent on species and stage of life cycle.

SPECIES SUMMARIES

A discussion of critical habitats (structural and water quality) of each selected species are summarized in the following paragraphs and are presented in tabular format at the end of this section.

Tarpon

Tarpon is an important recreational species in the Tampa Bay area. Very little information was available to assess critical habitats of tarpon in Tampa Bay. Most information on juveniles and adults was obtained from research in other Florida estuaries. Tarpon spawn in Gulf of Mexico waters offshore of Tampa Bay and their eggs and larvae are transported into the estuary by tides and currents. Juvenile tarpon prefer backwater mangrove and marsh areas with soft muddy bottom sediments and little or no submerged vegetation. Adults prefer a wide range of habitats including, lagoons, ditches, canals, tidal tributaries and the main bay. Juveniles and adults are euryhaline and are also tolerant of low dissolved oxygen conditions. It appears that minimum water temperatures may be an important factor influencing their distribution.

Bay Anchovy

The bay anchovy has a ubiquitous distribution and is one of the most common species in the Tampa Bay estuary. It is an important forage fish for many of the commercially and recreationally important species within the Bay. This species is quite tolerant of fluctuations in water temperature and salinity. Egg and larval bay anchovies prefer dissolved oxygen levels greater than 2.5-3.0 mg/L. The bay anchovy prefers shallow waters of the estuary; however, it does not appear to be associated with any type of submerged aquatic vegetation or structural habitat.

Striped Killifish

The striped killifish (sometimes referred to as longnose killifish) is distributed throughout much of the Tampa Bay estuary. Although this species is tolerant of extreme variations in water quality parameters such as salinity, water temperature and dissolved oxygen, it prefers higher salinity waters located along the shallow shorelines of Tampa Bay. Striped killifish distribution apparently is not influenced by the presence of submerged aquatic vegetation as it is commonly collected in both vegetated and non-vegetated habitats within the estuary.

Snook

Snook is an important recreational fish species in the Tampa Bay estuary. Snook spend the majority of their life cycle within and near the mouths of estuaries. Studies suggest that salinity may not be a major factor influencing larval and juvenile snook distribution in the estuary; however, water temperature, water depth, currents and structural habitat components appear to be critical factors in the selection of snook spawning and nursery habitat. Extremely low water temperatures can be lethal to snook. Snook abundance is probably limited by the availability of critical habitat in the Tampa Bay estuary. Typical habitats of larval, juvenile and adult snook include mangroves, backwaters, tidal tributaries and areas which provide access to deep water channels.

Spotted Seatrout

Spotted seatrout is an economically and ecologically important species in the Tampa Bay estuary. It supports valuable commercial and recreational fisheries. Spotted seatrout remain in the Tampa Bay estuary throughout their life cycle. It is tolerant of a broad range of salinities; however, it prefers poly- and mesohaline waters. Spotted seatrout is intolerant of extreme cold water conditions and may move into deeper, more thermally stable waters of the estuary to avoid rapidly changing temperatures. Although postlarvae and juvenile seatrout sometimes utilize non-vegetated backwater areas as nursery habitat, their critical habitat appears to be seagrass areas. Declines in seatrout abundance have been linked with the disappearance of seagrasses in the Tampa Bay estuary.

Red Drum

Red drum is an important recreational species in the Tampa Bay estuary. It is an estuarine dependent species which may spend part or all of its life cycle within the estuary. Preference for specific salinity regimes varies with red drum life stage. It appears that the interaction of water temperature and salinity are important to the survival and growth of eggs and larvae. In laboratory experiments, minimum dissolved oxygen requirements of juveniles were estimated to be approximately 2 mg/L. Red drum use a variety of habitats within the Tampa Bay estuary depending on life stage. Larvae are typically located in the deeper mid-bay regions of the estuary. Juveniles are found in both seagrass and non-vegetated backwater areas; however, critical nursery areas appear to be the non-vegetated oligohaline habitats. These areas are often characterized by the presence of marsh or mangrove vegetation growing along the shoreline. Critical habitats of adult red drum in the Tampa Bay estuary have not been adequately described.

Silver Perch

Silver perch is one of the most abundant fish species in the Tampa Bay estuary. Distribution and abundance of silver perch may be influenced by a variety of water quality and structural habitat parameters. Silver perch is euryhaline, but prefers meso- and polyhaline waters. It is tolerant of most typical warm water temperatures within the estuary; however, extreme low water temperatures may be lethal. Larval, juvenile and adult silver perch prefer habitats characterized by structure such as submerged aquatic vegetation.

spot

Spot is one of the most common fish species in the Tampa Bay estuary. Depending upon its life stage it may occur in all regions of the estuary. Spot is euryhaline and eurythermal and is tolerant of a wide range of environmental conditions. Spot can tolerate dissolved oxygen concentrations as low as 2 mg/L, although it is more common in waters with D.O. greater than 4 mg/L. Spot show little preference for specific habitats, although larvae and juveniles are typically associated with areas of structure such as seagrass, rocks, or seawalls. It is a benthic feeder and prefers areas with mud or mud-sand mixtures of sediment. Spot are known to occupy environmentally stressed regions of Tampa Bay.

Striped Mullet

Striped mullet is the most important inshore commercial finfish in Florida; however, very little research has been conducted on identifying its critical habitats. Salinity requirements of striped mullet are variable and dependent upon life stage. It appears that tidal tributaries in Tampa Bay are important nursery areas for juvenile mullet, and their distribution in these habitats may be contingent on salinity. Laboratory studies suggest that egg and larval striped mullet are sensitive to low dissolved oxygen conditions; however, no data were available for juveniles and adults. Larvae, juveniles and adults prefer shallow habitats in the Tampa Bay estuary. Juveniles and adults are typically found in areas with mud or mud-sand bottom substrates, with little or no submerged aquatic vegetation, in the main portions of the estuary and tidal tributaries.

Clown Goby

The clown goby is an ecologically important fish species in the Tampa Bay estuary. This species remains in the estuary during its entire life cycle. Little information was available to document critical habitats of the clown goby. This species tolerates a wide range of salinities and water temperatures in the Tampa Bay estuary. Juveniles and adult clown gobies prefer areas of meso- and polyhaline salinities and shallow portions of the estuary characterized by submerged aquatic vegetation.

Lined Sole and Hogchoker

Limited information was available on the critical habitats of lined sole and hogchoker in the Tampa Bay estuary. Both species are euryhaline, although the hogchoker prefers waters with salinities in the oligohaline to freshwater range and is typically found in tidal tributaries. Both species are commonly collected in shallow water regions; however, their distribution in deeper portions of the Bay have not been adequately described. Both species are found over vegetated and nonvegetated bottoms. Hogchoker appears to prefer nonvegetated bottoms in low salinity portions of tidal tributaries. Juvenile and adult lined sole may prefer seagrass areas to nonvegetated habitats.

Blue Crab

The blue crab is both commercially and recreationally important to Tampa Bay. The species is found throughout the estuary from the oligohaline tidal streams to the marine waters near the mouth. Environmental requirements which affect blue crab growth, survival, and distribution vary depending on life stage and sex. Blue crab eggs and larvae are most sensitive to fluctuations in environmental conditions such as temperature and salinity. Juvenile and adult crabs have a greater tolerance to these fluctuations and, in being very mobile, can avoid degraded areas when possible. Seagrass beds and other vegetated areas are important habitats for juvenile blue crabs in Tampa Bay. It appears that salinity rather than substrate type or benthic vegetation is a major factor influencing large juvenile and adult blue crab distribution in Tampa Bay.

Pink Shrimp

Pink shrimp support a sizeable bait-shrimp fishery in Tampa Bay and is an important link in the marine food web. Pink shrimp use the bay as a nursery area for about two to six months and when mature, move offshore to spawn. It appears that pink shrimp are capable of tolerating a wide range of temperatures and salinites, especially the upper range limits. Juvenile and adult pink shrimp have been documented in water temperatures to 38 °C and salinites to 45 ppt. Salt marshes, mangroves, and especially seagrasses are important nursery habitat for juvenile pink shrimp in Tampa Bay. These areas provide both protection and a food source. In Tampa Bay and other Florida waters, the loss of seagrass has been related to the decline of the bait-shrimp fishery.

American Oyster

Historically, the oyster was a commercially valuable species in the Tampa Bay estuary; however, anthropogenic activities have reduced its abundance by contributing to habitat degredation. High coliform levels have led to prohibited harvest in most areas of the Bay. Oysters complete their entire life cycle within Tampa Bay. The early life stage appear to be the most suseptible to fluctuations in environmental conditions such as temperature and salinity. Adult oysters can tolerate extreme environmental changes for short periods of time. In Tampa Bay, oysters are found in shallow bays, mud flats, and sand bars, and other firm structures. Soft mud and shifting sand are the only substrates unsuitable for oyster communitites.

Hard Clam

Both the northern and southern quahog are present in varying abundances in Tampa Bay. Hard clam abundance has decreased in Tampa Bay as a result of anthropogenic activities such as dredge and fill operations. Many clam beds have been closed to harvest due to high coliform levels in the surrounding water. In general, the hard clam completes it's life cycle within the Tampa Bay estuarine system. As with most estuarine organisms, hard clams are most susceptible to changes in environmental parameters during their early stages of development. Tolerance to parameters such as temperature and salinity increase as the clams reach the adult stage. This is primarily a function of attaining the ability to escape adverse stress by valve closure. Bottom substrate appears to be the primary factor responsible for the settling of hard clam larvae. Shelly substrates covered with a thin layer of detritus attract the largest sets of hard clams, followed by sandy sediments, with mud being the least colonized sediment. Adult hard clams are capable of living in a variety of sediment types; northern quahogs seem to prefer sand over mud, and associations between the southern quahog and seagrass have been reported; however, seagrass is apparently not essential for the survival of clams.

Grass Shrimp

Grass shrimp are important ecologically to Tampa Bay as they are a vital link in the marine food web. Four species of grass shrimp have been reported in Tampa Bay and they occur in varying abundances throughout the estuary. Grass shrimp can be considered a resident species because it completes it's life cycle within the Tampa Bay estuary. Temperature and salinity are considered to be the major factors limiting the distribution of grass shrimp. Tolerance to environmental conditions increases as grass shrimp progress through their life stages. Grass shrimp appear to be capable of tolerating a wide range of temperatures and salinities. They appear to prefer more turbid waters, possibly to avoid certain predators. Grass shrimp also use seagrass and other aquatic vegetation as a refuge from predation and for foraging.

Diopatra cuprea

The polychaete worm, *Diopatra cuprea* has been shown to be an important component of the Tampa Bay benthos by manufacturing tubes that provide important habitat for other benthic organisms. However, as with many bentic invertebrates, only limited information is available on this species; studies primarily address species distribution, feeding habits, and some environmental requirements as related to structural habitat. No life history information was available for this species.

Florida Manatee

The Florida manatee is found throughout Tampa Bay during the warm months, but is most abundant during the winter months near the warm water discharges of power plants. Three environmental requirements are critical for the survival of the Florida manatee in Tampa Bay; warm water in the winter months, fresh water to minimize osmotic stress and allow periodic drinking, and abundant seagrasses for food. Manatees prefer water temperatures above 20 °C temperatures below this can cause thermal stress and result in manatee mortalities if they are exposed for extended periods. Areas in Tampa Bay which provide critical habitat for the Florida manatee include the following: warm water outfalls at Bartow, Big Bend and Port Sutton power plants; Coffeepot Bayou; Hillsborough River; portions of the Little Manatee and Manatee Rivers; Braden River; Terra Ceia Bay; and Anna Maria Sound.

General and preferred ranges and upper and lower tolerance limits for environmental requirements of tarpon. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				26-30°C (E, L)	Wade 1962; Berrien et al. 1978; Smith 1980; Cyr 1991; Crabtree et al. in press
				17-33 (J)	Cyr 1991
			39 °C		Moffett and Randall 1957
		10 (J)			Robins 1978
		11 (J)			Tabb 1962
		12 (J)			Rickards 1968
Salinity (ppt)				28.5-39 (E, L)	Zale and Merrifield 1989
				0-47	Zale and Merrifield 1989
Dissolved Oxygen (mg/l)	tolerant of very low DO waters, potentially anoxic (J)				Wade 1962; Ellis 1956
Depth (m)	<2m (J)				Wade 1962; Rickards 1968; Cyr 1991
				in surface waters at depths to 1400 + (A)	Cyr 1991
				in surface waters at depths to 1400 + (A)	Cyr 1991
Substrate	soft mud, unvegetated bottoms, often mangrove areas (J)				Wade 1962, 1963; Harrington 1966; Rickards 1968, Cyr 1991

General and preferred ranges and upper and lower tolerance limits for environmental requirements of bay anchovy. Letters in parentheses indicate life stage. S = spawning, E = egg, L = larval, J = juvenile, **A** = adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)	>20 (E,L)				Phillips 1981
				10.8-32.5 **(J,A)**	Springer and Woodburn 1960
	24.5-32.5 **(J,A)**		33 (avoidance)		Galloway and Strawn 1975
			35-36		Chung and Strawn 1982
Salinity (ppt)				0-45	Robinette 1983
				0.3-36.1 **(J,A)**	FIMP unpubl. data
	>18 **(L)**				Peebles et al. 1992
	0.5-18 **(J,A)**				Haddad et al. 1990, 1992
Dissolved Oxygen (mg/l)				1.0 -15.2 **(J,A)**	FIMP unpubl. data
		3.0 **(E,L)**			Chesney and Houde 1989
Depth (m)	<2 m **(J,A)**				Springer and Woodburn 1960 Orth and Heck 1980; Gilmore 1987; Haddad et al. 1989, 1990, 1992; FIMP 1989, 1990
Substrate	None				Springer and Woodburn 1960 Orth and Heck 1980; Gilmore 1987; Haddad et al. 1992

General and preferred ranges and upper and lower tolerance limits for environmental requirements of striped killifish. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)		<9.6°C (J,A)			Rinckey and Salomen **1964**
				to 33°C (J,A)	Martin and Finucane **1968**
			34°C		Orr **1955**
Salinity (ppt)				to 76	Simpson and Gunter **1956**
				0-35 (J,A)	**FIMP** unpubl. data
	>25 (J,A)				**FIMP** unpubl. data
	>18 (J,A)				Haddad et al. **1992**
Dissolved Oxygen (mg/l)				1.4-11.1 (J,A)	**FIMP** unpubl. data
Depth (m)	<0.5 (J,A)				**FIMP 1989, 1990**
Substrate	sandy-mud (J,A)				Springer and Woodburn **1960**
	sandy-silt (J,A)				Martin and Finucane **1968**
				unvegetated to vegetated (J,A)	Haddad et al. **1992;** **FIMP 1989, 1990**

General and preferred ranges and upper and lower tolerance limits for environmental requirements of snook. Letters in

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)	27 (S)			13.1-35.6 (J)	Taylor et al. 1992
				13.1-35.6 (J)	McMichael et al. 1989, FIMP unpubl. data
		9.6, 11.8 (A)			Rinckey and Soloman 1964
		9.1-10.1 (A)			Taylor pers. comm. 1992
Salinity (ppt)				0-32 (J)	McMichael et al. 1989
				0-36 (J)	FIMP unpubl. data
				0-36 (A)	Thue et al. 1982
Dissolved Oxygen (mg/l)	4-7 (J)			1.3-8.4 (J)	Haddad et al. 1992
		0.4 (J)			Shafland and Koehl 1979
Depth (m)	Shallow shoreline where depth drops rapidly to 0.5 m (L,J)				Haddad et al. 1992
Substrate	mud, sand-mud (J)				McMichael et al. 1989
	Floating vegetation mangrove and structured shoreline (J)				McMichael et al. 1989
	red mangrove prop roots and muddy bottoms (L,J)				McMichael et al. 1989 Peters et al. 1992

in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				21-28 (S)	Kostecke 1984
	28 (E,L)			23-32.7 (E,L)	Taniguchi 1980
				14.1-29.7 (J)	FIMP (unpubl. data)
	16-27 **(A)**	7-10 **(A)**			Tabb 1966
		4-7 (A)			Gunter 1941; Gunter and Hildebrand 1951; Tabb 1958; Moore 1976
Salinity (ppt)	20-35 (S)				Arnold et al. 1976
	28.1 **(E,L)**			18.6-37.5 (E,L)	Taniguchi 1980
				8-40 (L)	Rutherford et al. 1986
				0-35 **(J)**	McMichael and Peters 1989
	>5 **(J)**				Peebles et al. 1992, Haddad et al. 1992
				0.2-70 (A)	Simmons 1957
	20 **(A)**	10 (A)	45 **(A)**		Wakeman 1978
		<5 (A)			Kostecki 1984
Depth (m)	**<2 (J)**				McMichael and Peters 1989; Edwards 1990; Peters et al. 1992
Substrate	seagrass **(L,J,A)**				Springer and Woodburn 1960; McMichael and Peters 1989; Rutherford et al. 1989
				unvegetated - vegetated	Lorio and Perret 1980; McMichael and Peters 1989

General and preferred ranges and upper and lower tolerance limits for environmental requirements of red drum. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)	25-30 (E,L)			20-30 (E,L)	Holt et al. 1981
				12.5-32.2 (J)	Peters and McMichael 1987 FIMP. (unpubl. data)
				2-33 (J)	Gunter and Hildebrand 1951 Simmons and Breuer 1962
Salinity (ppt)	39 (E,L)			15-30 (E,L)	Holt et al. 1981
				16-34 (L)	Peters and McMichael 1987
				8-35 (L)	Rutherford et al. 1986; Haddad et al. 1992
				0-37 (J)	FIMP (unpubl. data)
				2-40 (J)	Rutherford et al. 1986
				0-21 (J)	Edwards 1990
D.O. (mg/l)		2 (J)			Neill 1990
Depth (m)	variable (E,L)				Robinson 1985; Holt et al. 1983; Peters and McMichael 1987
	0.25-2 (J)				Peters and McMichael 1987
Substrate	seagrasses (L,J)				Holt et al. 1983
	seagrasses, backwaters, muddy bottoms (L,J)				Peters and McMichaels 1987; Springer and Woodburn 1960; Edwards 1990

General and preferred ranges and upper and lower tolerance limits for environmental requirements of silver perch. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)		13°		10-32.5°C	Springer and Woodburn 1960
			34-37 (3-hr LD_{50})		Chung 1977
			37-40 (30 min LD_{100})		Chung 1977
Salinity (ppt)				1-32 (L)	Peters and McMichael unpubl. manuscript
	>10 (L)				Peters and McMichael unpubl. manuscript
	>20 (J)				Springer and Woodburn 1960
Dissolved Oxygen (mg/L)				1.3-15.2	FlMP unpubl. data
Depth (m)	<2 (J)				FlMP 1989, 1990
				to 5-10	Moe and Martin 1965
Substrate	seagrass, rocks piers, seawalls (L)				Peters and McMichael unpubl. manuscript
	seagrass (J)				Peters and McMichael unpubl. manusript
				sandy, unvegetated	Moe and Martin 1965

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature ($^{\circ}$C)				1.2-35.5	Hedgepath 1967; Parker 1971; Moser and Gerry 1989
	25-34°C				Gallowav and Strawn 1974
		14 (E)	28(E)		Hettler and Clements 1978
				6-20 (J)	Parker 1971
		10 (A,L)			Parker 1971; Hoss et al. 1981
		4-5			Dawson 1958
			31-38 (J)		Bridges 1971; Horton and Bridges 1973; Hoss et al. 1972, 1973
			35-36 3 hr LD_{50} (J)		Chung 1977
			36-38 30 min LD_{50} (J)		Chung 1977
Salinity (ppt)				0-60	Hedgepath 1967; Johnson 1978
				6-32 (L)	Peters and McMichael, unpublished ms.
				0-35 (J)	Peters and McMichael, unpublished ms.
	oligohaline to marine				Haddad et al. 1992; Homer and Mihursky 1991
Dissolved				to <2 (J,A)	Thorton 1975; Burton et al. 1980; Rothschild 1990
	>4 (J,A)				Markle 1976: Chao and Musick 1977: Rothschild 1990
			0.49 (J) 1 hr LC_{50}		Burton et al. 1980
			0.70 (J) 96 hr LC_{50}		Burton et al. 1980
Depth (m)	<2 (J)				Springer and Woodburn 1960; Peters and McMichael, unoublished ms.
	>2 (J,A)				Coastal Environmental Services 1992
				seasonal and dependent on life stage	Peters and McMichael, unpublished ms.
Substrate	structure-rocks, seagrass (L,J)				Peters and McMichael, unpublished ms.
	muddy sediments (J,A)				Homer and Mihursky 1991
				mud-sand (J,A)	FIMP 1989, 1990

General and preferred ranges and upper and lower tolerance limits for environmental requirements of striped mullet. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)	18.9-25.3 (E,L)			10.3-3.9 (E,L)	Nash and Sylvester 1974
				13-34.5 (A,J)	Kilby 1949
				10.7-32.5 (A,J)	Springer and Woodburn 1960
			37-38 3 hr-LD_{50} (J)		Chung 1977
			39-41 30 min-LD_{100} (J)		Chung 1977
		10 (A)			Gilmore et al. 1978
Salinity (ppt)	>32 (E)				Sylvester et al. 1975
	26 (L)				Sylvester et al. 1975
	30-40 (E)			10-55 (E)	Lee and Menu 1981
	0.5-18(J)				Haddad et al. 1992
				0-75	Collins 1981; Simmons 1957
				0.3-35(J)	FIMP unpubl. data
Dissolved Oxygen (mg/l)		4 (L)			Sylvester et al. 1975
		54.5 (E)			Sylvester et al. 1975
Depth (m)	surface waters (E,L)				Arnold and Thompson 1988
	< 2 m (J)				FIMP 1989, 1990
Substrate	mud, mud-sandy (J)				FIMP 1989, 1990; Haddad et al. 1992

General and preferred ranges and upper and lower tolerance limits for environmental requirements of clown goby. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, **J**=juvenile, **A**=adult.

				Range	Reference
Temperature				**12-34.1 (J,A)**	Darcy 1980
(°C)		10.8 "stunned"			
				14.8-31.6 **(J,A)**	FIMP unpubl. data
Salinity (ppt)				0-36.6 **(J,A)**	Darcy 1980
	20-30 **(J,A)**				Springer and Woodburn 1960
				to 44 **(J,A)**	Fonseca unpubl. data, 1992
				0.3-37 **(J,A)**	FIMP unpubl. data
Dissolved					
Oxygen (mg/l)					
Depth (m)	**<2 (J,A)**				Kilby 1955; Haddad et al. 1992
Substrate	seagrasses, muddy vegetated bottoms **(J A)**				Reid 1954; Haddad et al. 1992; Springer and Woodburn 1960; FIMP 1989, 1990
				Bare areas to dense vetegation **(J,A)**	Fonseca unpubl. data, 1992

General and preferred ranges and upper and lower tolerance limits for environmental requirements of lined sole. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				11.2-32.4 **(J,A)**	Springer and Woodburn 1960
	20 (S)				Futch 1970
			32 (L)		Houde 1974
Salinity (ppt)				4-35 **(J,A)**	Springer and Woodburn 1960
	>5 (E,L,J,A)				FIMP unpubl. data; FIMP 1989, 1990; Haddad et al. 1992; Peebles et al. 1992
Dissolved Oxygen **(mg/l)**					
Depth (m)	**<2 (J,A)**				Haddad et al. 1992
					FIMP unpubl. data
Substrate				vegetated-non-vegetated **(J,A)**	Haddad et al. 1990, 1992

General and preferred ranges and upper and lower tolerance limits for environmental requirements of hogchoker.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				14.2-30.2 **(J,A)**	Futch 1970b; Houde 1974; FIMP unpubl. data
Salinity (ppt)	**> 5 (L)**				Springer and Woodburn 1960
	0-18 **(J)**			**0-36** **(J,A)**	FIMP unpubl. data
Dissolved Oxygen (mg/l)					
Depth (m)				all depths **(J,A)**	Haddad et al. 1992; FIMP 1989, 1990; Coastal Env. Ser. 1992
Substrate				vegetated - non-vegetated **(J,A)**	Haddad et al. 1990, 1992; FIMP 1989, 1990
	non-vegetated **(J,A)**				Haddad et al. 1990, 1992; FIMP 1989, 1990

General and preferred ranges and upper and lower tolerance limits for environmental requirements of blue crab. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)	19-29 (E)				Sandoz and Rogers 1944
	25 **(L,** at 30 ppt)				Costlow and Bookhout 1959; Sulkin and Epifanio 1975; Bookhout et al. 1976
		21 (L)	**30 (L)**		Williams 1965
			33 (J)		Holland et al. 1971
		5.5 (J)			Van Heukelen and Sulkin 1990
				variable depending on salinity and sex	Steele and Bert (unpubl. manuscript)
	23-33 (E)				Sandoz and Rogers 1944
	30 (L at 25°C)				Costlow and Bookhout 1959; Sulkin and Epifaunio 1975; Bookhout et al. 1976
Salinity (ppt)	22-28(E)				Newcombe 1945
				2-21 (J)	Holland et al. 1971
				variable depending on age and sex	Steele and Bert (unpubl. ms)
D.O. (mg/l)		**<0.5 (J,A)**			Lowery and Tate 1986
Substrate	seagrass **(L)**				Lipcius et al. 1990; Steele pers. comm. 1992
				seagrass, unvegetated, macroalgae (A)	Steele pers. comm. 1992; Heck and Thoman 1984; Williams 1984

General and preferred ranges and upper and lower tolerance limits for environmental requirements of oyster. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)	20-30 (A)				Stanley and Sellers 1986
	30-32.5 (L)				Davis and Calabrese 1979
		15 (L)	35 (L)		MacInnes and Calabrese 1964
			35 (A)		Quick 1971; Tinsman and Mauer 1974
Salinity (ppt)	10-30 (L)			5-35 (L)	Carriker 1951, Davis 1958, Calabrese and Davis 1970
				16-30 (L)	Finucane and Campbell 1969
	10-30 (A)			5-40 (A)	Butler 1954, Gunter and Geyer 1955, Galtsoff 1964, Stenzel 1971
				5-30 (A)	Dawson 1953
		<2 (A)			Gunter 1953
D.O. (Mg/L)				to <1 (L,A)	Sparks et al. 1958, Widdows Et al. 1989
Currents (cm/sec)	156-200+ (L,J,A)				Veal et al. 1972
				11-66 (L,J,A)	Wells 1961
			150 (L,J,A)		Mackenzie 1981
Depth (m)				0.3-5 (L,J.A)	Dawson 1973
				0.3-12 (L,J,A)	Butler 1954
pH	7.8-8.2 (S)	6 (S)	10 (S)		Calabrese and Davis 1969
	6.75-8.75 (L)	6-5 (L)	9 (L)		Calabrese and Davis 1966
Substrate	firm shell, rock (L,J,A)				Kennedy 1991
				mud flats, sandy areas,. hard bottoms (L,J,A)	Kennedy 1991

General and preferred ranges and upper and lower tolerance limits for environmental requirements of hard clam. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower **Limit**	Upper **Limit**	Range	Reference
Temperature (°C)				**18.5-28.3**(S)	Hesselman et al. **1989**
		10 (L)			Davis and Calabrese **1964**
	Dependent on salinity (L)				Davis and Calabrese **1964**
	20 (A)	**9 (A)**	**31** (A)		Ansell **1967**
		10-12 (A)			Menzel **1961, 1962**
			35 southern (A)		Mulholland **1984**
			30 (A)		Arnold et al. **1991**
Salinity (ppt)	**26.5-27.5(E)**			**20-35 (E)**	Davis **1958**
		15 (L)	**35 (L)**		Loosanoff and Davis **1963**
		17.5 (L,J)			Castagna and Chanley **1973**
	35-36 southern **(A)**	**20** (A)			Woodburn **1961, 1962**
	24-28 northern (A)	**12.5**(A)			Castagna and Chanley **1973**
	> 24 southern (A)				Taylor and Saloman **1970**
Dissolved Oxygen (mg/l)	**>4.2** northern (L)	**2.4**(L)			Morrison **1971**
		to **0.5**(L)			Morrison **1971**
	75 northern **(A)**	to 1 (A)			Savage **1976**
				4-7.8southern (A)	Godcharles and Jaap **1973**
pH	**7.5-8**northern (L)			**6.75-8.5**(L)	Calabrese and Davis **1966**
	r 7 northern (A)		**9** (A)		Calabrase **1972**
Current (cm/s)	**12-130 (L)**				Carriker **1952**
Depth (m)				<5 northern (A)	Godcharles and Jaap **1973**
				4.7-9.2 southern (L,J,A)	Mullholland **1984**
Substrate	shelly, thin layer detritus (L)			sandy (L), mud bottoms	Carriker **1961;** Keck et al. **1974**
		silty (A)			Pratt and Campbell **1956**
	seagrass southern (A)				Simms and Stokes **1967;** Taylor and Saloman **1968, 1970;** Godcharles **1971**

General and preferred ranges and upper and lower tolerance limits for environmental requirements of grass shrimp ***(P. pugio).*** Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	**Lower Limit**	**Upper Limit**	**Range**	**Reference**
Temperature (°C)				5-38 (L,A)	Wood 1967, Christmas and Langley 1973
	18-25 (A)	5 **(A)**	38 (A)		Wood 1967
	18-20 (S)				Wood 1967
	20-30 **(L)**	15 **(L)**	35 (L)		McKenney and Neff 1979; Sandifer 1973
Salinity (ppt)	20-25 **(L)**	3 **(L)**	31 (L)		Floyd 1977; McKenney and Neff 1979; Knowlton and Kirby 1984
		16 (L) LD_{50}	**46 (L) LD_{50}**		Kirby and Knowlton 1976
		3 (L) LD_{50}			McKenney and Neff 1979
	4-16 (A)			1-55 (A)	Wood 1967; Swingle 1971; Bowler and Serdenberg 1971; Christmas and Langley 1973; Kirby and Knowlton 1976; Morgan 1980
Dissolved Oxygen **(mg/l)**				2.8-4.4 (A)	Rozas and Hackney 1984
				6-11 (A)	Barrett et al. 1978
Depth **(m)**				to 16 (J,A)	Williams 1965
Substrate	aquatic vegetation - seagrasses **(J,A)**				Thorp 1976; Morgan 1980; Coen et al. 1981; Heck and Thoman 1981; Knieb 1987; Livingston et al. 1976

General and preferred ranges and upper and lower tolerance limits for environmental requirements of *Diopatra cuprea.* Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature				9-29 (A)	Simon and Dauer 1977
(°C)		1.8 (A)			Myers 1972
Salinity (ppt)				22.3-30.1 (A)	Bloom et al. 1972; Bell and Coen 1982
Current (m/s)				.09-.6 (A)	Mangum et al. 1968
Depth (m)				4-5 (A)	Santos and Simon 1980
				0.8-1 (A)	Bell and Coen 1982; Virnstein 1972
Substrate				sandy gravel to fine grained mud (A)	Mangum 1968, Bell and Coen 1982

General and preferred ranges and upper and lower tolerance limits for environmental requirements of juvenile and adult manatee. Letters in parentheses indicate life stage.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)	>20°C				O'Shea and Kochman 1990
		13.5°C			Hartman 1979
Salinity (ppt)	<25	0	33		Hartman 1979
				Freshwater to marine	Hanman 1979
Depth (m)	1.5-2 m	.7			Hartman 1979; FDNR, unpublished data
Substrate	Seagrasses or other aquatic vegetation			Various	Reynolds and Odell 1991; Hartman 1979; FDNR, unpublished data

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ABBREVIATIONS

CCMP = Comprehensive Conservation and Management Plan

DO = Dissolved oxygen

FDNR = Florida Department of Natural Resources

FMRI = Florida Marine Research Institute

GCRL = Gulf Coast Research Lab (Biloxi, Mississippi)

HCEPC = Hillsborough County Environmental Protection Commission

LC_{50} = Lethal concentration (50%)

LD_{50} = Lethal dose (50%)

m = Meters

mg/L = Milligrams per liter

NEP = National Estuary Program

PCB = Polychlorinated biphenyl

PPt = Parts per thousand

rkm = River kilometer

SAV = Submerged aquatic vegetation

SL = Standard length

TL = Total length

USF = University of South Florida

YOY = Young of year

1.0 INTRODUCTION

1.1 OBJECTIVES

The National Estuary Program (NEP) was established by the Water Quality Act of 1987, to protect and improve the water quality and ecological integrity of nationally significant estuaries. Tampa Bay was accepted into the NEP in 1990. One of the purposes of the Tampa Bay National Estuary Program (TBNEP) is to identify environmental problems within the estuary and to assess the status of natural resources which may be impacted by those problems. The results of the research will be used to develop a Comprehensive Conservation and Management Plan (CCMP) directed at restoring and protecting the estuary.

A comprehensive evaluation and synthesis of critical habitats of fish and wildlife species is lacking in previous reports on the Tampa Bay estuary. The purpose of this report is to characterize habitat types critical for selected species indigenous to Tampa Bay. The results of this project will be used to guide the development of the most effective strategies for habitat restoration and protection and to identify information necessary for future management decisions.

The first objective of this project was to identify important biological resources in the Tampa Bay estuary. The subtropical location of Tampa Bay is in part responsible for a large diversity of organisms which contribute to a complex ecosystem. Over 200 species of finfish (Comp 1985) and numerous other fauna comprise the Tampa Bay ecosystem. Although many of these species can be considered important to the estuary, or are dependent upon the estuary, a selection process must be used to identify those species most important to the management goals of the TBNEP.

Two ecosystem attributes of great importance to society are biotic integrity of the ecosystem and resources of direct use to humans (U.S. EPA 1990; Frithsen et al. 1991b). Therefore, both scientific and human use values must take into account the selection of important biological species. Measures of biotic integrity generally focus upon the presence of indigenous species, and the health or condition of individuals within each population. Human use attributes relate directly to issues of public concern. For example, the public values the ability of estuarine waters to support commercial and recreational fishing. The aesthetic value of certain species are another important consideration. These species are important not because of the ecological roles they might play or their economic importance, but because their presence is valued by the general public and various users of the estuary. In the Tampa Bay estuary, this would include such species as the manatee, bottlenose dolphin and many bird species. Therefore, the selection of important fish and wildlife species in the Tampa Bay estuary should use criteria relating to both biotic integrity and resource use. We chose to identify important species in the Tampa Bay estuary using the following criteria:

- Ecological importance

- Commercial and recreational importance
- Aesthetic importance
- Status as endangered, protected or rare species

These four criteria are not mutually exclusive and many species can be rated using more than one selection criteria. Similar guidelines were used to identify key biological resources in the Chesapeake Bay (Richkus et al. 1991; Funderburk et al. 1991) and the Delaware Estuary (Frithsen et al. 1991a) and were found to be an effective way to focus efforts to assess status and trends in abundance and to evaluate critical habitats.

Selection of ecologically important species is difficult because specific ecologic roles of many species have not been fully determined. However, the data that exist support the inclusion of this criteria in the assessment of important fish and wildlife species. For example, many abundant fish species, including anchovies ***(Anchoa mitchilli),*** silversides ***(Menidia*** spp.) and gobies ***(Gobiosoma*** spp. and ***Microgobius*** spp.) are important prey for commercial and recreational fish species such as spotted seatrout (McMichael and Peters 1989).

It is easier to identify those species that are of commercial or recreational importance, or are considered aesthetically important. These species have typically received more attention because changes and trends in their abundance are of great interest to the public and of major concern to agencies responsible for managing natural resources.

Endangered species were included in the selection criteria because these species may provide valuable insight into the determination of critical habitats. The greatest cause of declines in diversity and abundance in terrestrial species has been identified as habitat decline. It is likely that similar declines in estuarine-dependent and wetland species are also correlated with habitat declines.

The second objective of this project was to identify habitat types critical to the chosen species. Habitat plays an important critical role in defining the success of any given species within a system. Habitat refers to the specific structural, physical, and chemical environment in which an organism lives (Haddad 1989).

Many of the species in Tampa Bay are dependent on the estuary for nursery, protection and foraging areas. Harris et al. (1983) estimated that over 70% of marine commercial and recreational fisheries are considered estuarine dependent.

The Tampa Bay estuary has been faced with extreme development pressures since the turn of the century, much of which has directly or indirectly caused significant declines in natural habitats. Lewis et al. (1985) estimated a 81% decline in seagrass meadows in Tampa Bay since the late 1800s. Large declines in saltmarsh (-30%), seagrass (-50%) and wetlands (-21%) occurred between the 1950s and 1982 (Haddad 1989).

In this report, we synthesize information collected on the life history and ecology of bay species and identify critical habitats potentially important to the sustenance of and ecological condition for those species.

1.2 METHODS

1.2.1 Selection of Important Biological Resources

The identification of biological resources for the Tampa Bay estuary required a thorough review of available information concerning the biology and ecology of the estuary. This involved computerized data base searches, and numerous contacts with federal, regional, state and university staff with expertise pertaining to the Tampa Bay estuary.

Additional literature was obtained by visiting local libraries in the Tampa Bay area and reviewing available journal articles and other documents. A bibliographic database of Tampa Bay references, compiled by the Center for Nearshore Marine Science at University of South Florida, was reviewed for pertinent information. Consultation with biologists at the Florida Department of Natural Resources Florida Marine Research Institute provided valuable insight into ongoing research and unpublished information. Fisheries independent datasets specific to Tampa Bay were obtained from FDNR's Fish Statistics Section and from the National Marine Fisheries Service.

Based upon this review, a preliminary list of important fisheries and wildlife species was produced. After consultation with members of the local scientific community and members of the TBNEP, **13** finfish, 5 invertebrate, **1** marine mammal, and soft bottom benthic communities were chosen for the assessment of critical habitats (Table 1-11).

1.2.2 Data Synthesis and Presentation

Each chapter in the report summarizes information for a selected species. Finfish species will be presented first, followed by shellfish, benthic communities and marine mammals. Data specific to the Tampa Bay estuary was reported when available, although data from other geographic areas were often used to supplement this information. The format used in the presentation of each species chapter is as follows:

- Introduction - This section provides general information on geographic distribution of the species, pertinent systematic or taxonomic information, and discusses importance of the species to the estuary (e.g. economic, ecologic, endangered status).
- Life History - This section summarizes information on the reproductive biology (e.g. spawning location and season, fecundity, size at maturity), age and growth, and population dynamics (e.g. migrations, mortality) for a selected species. Information

Table 1-1. List of species chosen for synthesis of life history information

Fish	
Tarpon	*Megalops atlanticus*
Bay anchovy	*Anchoa mitchilli*
Striped killifish	*Fundulus majalis*
Snook	*Centropomus undecimalis*
Spotted seatrout	*Cynoscion nebulosus*
Red drum	*Sciaenops ocellatus*
Silver perch	*Bairdiella chrysoura*
spot	*Leiostomus xanthurus*
Striped mullet	*Mugil cephalus*
Clown goby	*Microgobius gulosus*
Lined sole and Hogchoker	*Achirus lineatus and Trinectes maculatus*
Invertebrates	
Blue crab	*Callinectes sapidus*
Pink shrimp	*Penaeus duorarum*
American oyster	*Crassostrea virginica*
Hard clam	*Mercenaria rnercenaria,* *M. campechiensus*
Grass shrimp	*Palaemonetes spp.*
Benthic Community	
	Diopatra cuprea
Marine Mammals	
Florida Manatee	*Trichechus manatus latirostris*

relating to each life stage were presented when possible. Knowledge of life history is essential in identifying life stages that may be particularly susceptible to declines or alterations in critical habitats.

- Ecological Role - This section is included to provide a means of assessing the ecologic position of this species in the Tampa Bay ecosystem. Information is presented on prey species at different life stages, and possible predators or competitors.

- Contaminants - This section addresses the issue of toxicants (trace metals, pesticides, PCBs) in fish and wildlife in the Tampa Bay estuary. Some chemicals, if they occur in sufficiently high concentrations, can cause adverse biological impacts on marine and estuarine organisms (Long et al. 1991). Information on toxicants relating to selected species are presented where data were available.

- Habitat Requirements - Habitat requirements for selected species involve the complex interaction of numerous physical, chemicat and biological interactions. Habitat types were divided into two categories; water quality; and, structural. Components of the water quality category included; salinity, water temperature, dissolved oxygen, turbidity, and pH. Structural habitat parameters included; vegetation, substrate, water depth, shoreline and basin morphology. A thorough understanding of habitat requirements are essential for management agencies responsible for maintaining or improving the status of a particular species.

Information on optimum or preferred as well as minimum or tolerable habitat conditions is presented when available. Optimum levels are important in setting habitat goals that will help restore a species to historic population levels. Defining minimum limits is important in deciding which species are particularly sensitive and need immediate attention (Funderburk et al. 1991)

2.0 TAMPA BAY PHYSICAL SETTING

To place species specific habitat requirements within a proper context, it is necessary to provide a brief description of the physical setting of the Tampa Bay estuary. Tampa Bay is Florida's largest (1030.8 km^2) open water estuary. It is located on the west coast of the Florida peninsula between latitude 27° 30' and 28° 00' (Fig. 2-1). The Tampa Bay watershed covers approximately 5,700 km^2 (Lewis and Estevez 1988).

Unlike many estuaries in the United States, Tampa Bay is not associated with a single large river as are many east coast estuaries. Four natural rivers (Hillsborough, Alafia, Little Manatee and Manatee) flow into Tampa Bay (Fig. 2-1). All originate on the Florida peninsula and consequently, are relatively small (Flannery 1989). These rivers all rise to the east of Tampa Bay and flow 65-80 km southwest or west (Lewis and Estevez 1988). Despite their limited size, tributaries to Tampa Bay are important influences on its physio-chemical characteristics.

Tampa Bay is classified as a subtropical estuary that is characterized by long, warm, humid summers and warm winters (Clark and MacAuley 1989). The average bay area temperature is 23°C and freezing temperatures are experienced only four nights each year on average.

Tampa Bay is considered a vertically well-mixed and generally unstratified estuary (Lewis and Estevez 1988). The predominant tidal cycle in Tampa Bay is semidiurnal (two high and two low daily). Tidal heights range between 0.60 and 0.85 m (HCEPC 1984). Maximum current velocities generally occur near the mouth of the bay and may range from 1.8 m/sec on ebb tides to less than 1.1m/sec on flood tides. In upper Hillsborough and Old Tampa bays, current velocities decrease to as little as 10% of those at the bay mouth (Drew 1990).

Tampa Bay consists of open water and vegetated intertidal zones. Ninety-three percent of the bay is open water and 7% is vegetated intertidal area with mixtures of mangrove and tidal marsh vegetation (Lewis and Estevez 1988). Around the periphery of the bay there is a shallow shelf varying in width from 500 to 1,200 m, with maximum depth of approximately.1.5 m at its outer edge. Upon this submerged estuarine shelf grow the majority of the algae and seagrasses in the bay. Outside of the shelf, the bay drops off to natural depths of 7 m (Lewis and Estevez 1988).

The Tampa Bay estuary has historically been an area of rapid growth and development. During the last century, population estimates in the Tampa Bay area have increased from approximately 87,923 people (1910: U.S. Department of Commerce 1913) to 2.06 million people (1987: BEBR 1988, cited from Clark and MacAuley 1989). Population projections for the Tampa Bay area suggest that population density during the period 1988 to 2010 will increase by at least 15% and that by the year 2010, Pinellas County will be the most densely populated coastal county in the Gulf of Mexico (Culliton et al. 1990).

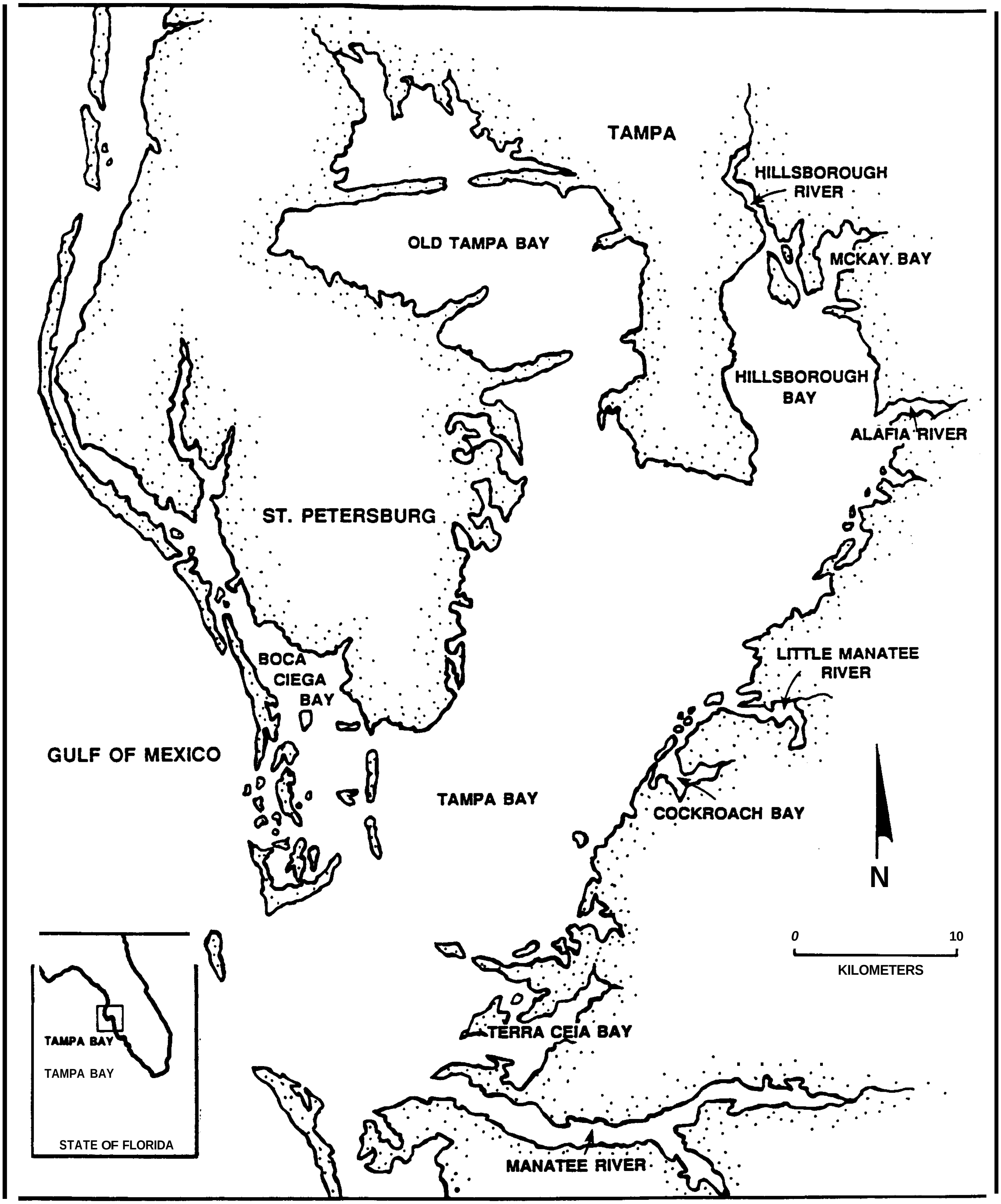

Figure 2-1. Diagram of the Tampa Bay estuary

The rapid growth and development of the Tampa Bay estuary region has been linked to declines in the areal extent and quality of specific habitats important to sustaining local species populations. For example, loss of habitat has been linked with dredge and fill activities, declining water quality, and ditching and diking wetlands (Lewis et al. 1985; 1989). Continued declines in habitat and biological resources within the estuary contributed to its inclusion into the National Estuary Program.

3.0 FISH

3.1 TARPON *(Megalops at/anticus)*

3.1.1 INTRODUCTION

The tarpon is found in tropical, subtropical, and temperate coastal waters of the Atlantic Ocean and Gulf of Mexico. In the western Atlantic, tarpon have been reported as far north as Nova Scotia, however, it is common from Virginia southward along the Atlantic coast, in the Florida Keys, the Gulf of Mexico and south to Brazil. Their primary center of abundance occurs around Florida, the Gulf of Mexico, and Central and South America (Wade 1962, 1969; Hildebrand 1939; Nelson 1984).

Tarpon belong to the monogeneric family Megalopidae, which includes only one other species. Phylogenetically, they are considered an ancestral species and they are located near the base of the teleostean classification (Gosline 1971). The presence of a leptocephalus larval stage is one character which contributes to this ancestral classification.

Tarpon are extremely valuable to the economies of coastal regions where they are fished (Cyr 1991). They are considered the premier inshore big-game fish of the Florida coast, primarily as a result of their large size (up to 300 pounds) and fighting ability (Robins 1978; Schomer and Johnson 1990). The tarpon fishery is worth millions of dollars to Florida's economy and no doubt exceeds a million dollars in the Boca Grande region alone (Wade and Robins 1973). To monitor the recreational tarpon fishery, the State of Florida recently established a $50 fee for each tarpon harvested. No commercial tarpon fisheries exist in the United States because the flesh is considered inedible (Zale and Merrifield 1989).

3.1.2 LIFE HISTORY

Spawning season and location of spawning have been examined along the west coast of Florida. Cyr (1991) examined reproductive condition of a total of 209 adult tarpon collected near Boca Grande (n=141), Tampa Bay (n=34) and other south Florida areas. Prespawn female tarpon were captured every month from March to July. Approximately 80% of the females were at peak reproductive development during May and June. Spent females were rare in the spring and summer and comprised 3.7%, 13.6%, and 25% of the females in the months of May, June and July, respectively. In August, the percentage of spent females rose to 89%. Ripe males occurred only in May, June and July and made up 7.4%, 28% and 29% of the male catch, respectively. Spent males were also captured in May, June and July and made up 7.4%, 4% and 20% of the male catch, respectively (Cyr 1991).

Many studies have reported that tarpon spawn offshore in both the Gulf of Mexico and Atlantic Ocean (Wade 1962; Hildebrand 1963a; Eldred 1967 1968; Cyr 1991; Crabtree et al., in press). Crabtree et al. (in press) suggested that adults undergo a spawning migration from inshore feeding areas to offshore spawning grounds. Tarpon appear to form pre-spawning aggregations prior to offshore spawning migrations and have been observed in large schools (25-200 individuals) 2 to 5 km offshore the west coast of Florida during late spring and summer.

The collection of larval tarpon in offshore waters supports the theory of offshore spawning. Cyr (1991) and Crabtree et al. (in press) collected larval tarpon at depths of 0 to 20 meters in 90 to 1400 meters of water (Fig. 3-1). These authors collected 54 larvae (7.3 to 24.4 mm) in 1981 and 275 larvae (3.5 to 24.4 mm) in 1989 in these offshore areas. Back-calculated hatch dates predicted from larval lengths suggested that these fish were spawned from May 20 to July 10. Additional collections would be necessary to delineate the entire spawning season, although in Florida it probably occurs from April through September (Cyr 1991; Crabtree et al. in press). Reproductive activity of tarpon appears to be strongly seasonal, and during the summer months tarpon larvae may be among the most common leptocephali in the waters over the outer continental shelf and slope in the eastern Gulf of Mexico (Crabtree et al. in press).

Leptocephalis tarpon larvae probably enter the Tampa Bay estuary using tides and currents. Although little information is available on Tampa Bay specifically, juvenile inhabit mangrove-marsh areas with soft muddy bottom sediments and no submerged vegetation (Harrington 1966; Richards 1968; Cyr 1991). Adult tarpon are considered to be euryhaline and are often collected in lagoons, ditches, canals, rivers, and estuaries (Wade 1962). More research is needed to document additional life history characteristics of tarpon in Tampa Bay.

Cyr (1991) summarized general age and growth parameters of juvenile and adult tarpon. Growth rates of young of the year tarpon are extremely seasonal, with exponential growth occurring from March to September and asymptotic growth during the winter months (Cyr 1991). Tarpon are a sexually dimorphic species, with females attaining a significantly greater size than males. Growth in length of both sexes approaches an asymptote; however, growth in weight may continue in females after growth in length ceases (Cyr 1991). Tarpon appear to mature at a relatively large size and late age. Cyr (1991) reported that the smallest mature female he collected was 1245 mm and approximately 13.8 years old. The smallest mature male was 1181 mm; however, no age was estimated for this fish. Cyr (1991) emphasized the need for more research in this area. Tarpon are extremely long-lived fish and ages exceeding 50 years have been reported in captive fish (Cyr 1991).

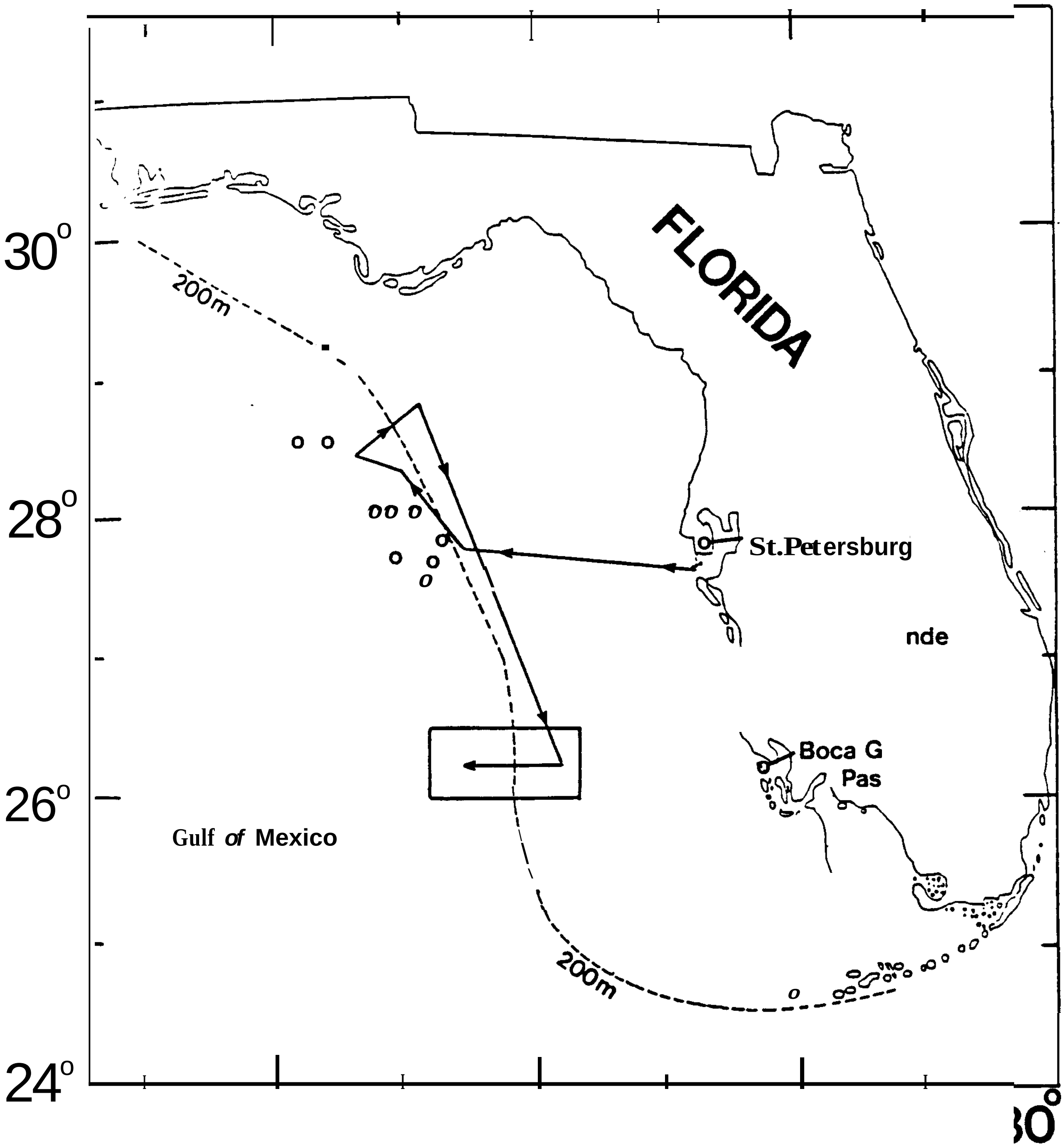

Figure 3-1. Map of Florida and surrounding waters showing tarpon *(M. atlanticus)* collection sites of 1981 and 1969 cruises. Solid line is 1989 cruise track; square is area in which 99% of larvae were collected in 1989. Circles depict locations in which larvae were collected during the 1981 cruise. Continuous dashed line is 100 fm curve. Source: Cyr 1991.

3.1.2.1 Ecological Role

No data were available to describe feeding habits of tarpon in the Tampa Bay estuary; however, this information was available from other estuaries. Postlarval tarpon taken from tidal creeks and pools near Sapelo Island, Georgia were strictly carnivorous and predominantly piscivorous (Rickards 1968). The principal food varied with relative availability of different food organisms and the size of the food consumed was directly related to the size of the tarpon. More advanced larval and early juvenile tarpon (less than 125 mm SL) feed primarily on zooplankton, including copepods and ostracods and secondarily on insects and small fishes. Larger juveniles continue to feed on zooplankton but progressively increase their consumption of insects, fishes, (especially poecillids and cyprinodontids) crabs, and grass shrimps of the genus ***Palaemonetes*** (Beebe 1927; Breder 1933; Moffett and Randall 1957; Harrington and Harrington 1960 1961; Rickards 1968; cited from Zale and Werrifield 1989). Adult tarpon are strictly carnivorous and feed primarily on mid-water prey (Hildebrand 1963a). Adult tarpon are known to prey upon mullet, pinfish, sunfish, sea catfish, sardines, needlefish and crustaceans (Wade 1962).

3.1.2.2 Predators

Tarpon eggs are probably eaten by predaceous plankton and small fishes (Wade 1962). Rickards (1968) reported that no other fish in a Georgia salt marsh were large enough to consume young-of-the-year tarpon other than other tarpon; however, there was a potential for competition from other species of carnivorous fish such as ladyfish, ***Elops saurus*** and spotted seatrout, ***Cynoscion nebulosus.*** Piscivorous birds, including herons, pelicans, and kingfishers are potential predators of juvenile tarpon (Beebe 1927; cited from Rickards 1968). Sharks and alligators are among the few natural enemies of adult tarpon.

3.1.3 CONTAMINANTS

The tarpon is a long-lived fish which occupies a position at the top of the food chain in the Tampa Bay estuary. The combination of these factors could contribute to the accumulation of toxic contaminants, such as trace metals, PCBs, or DDT in their tissues. Because the tarpon is not considered a foodfish in the estuary, the principal concern is whether contaminant levels are high enough to impact tarpon survival and population size.

Currently, no data exists to address these concerns. Harrington and Bidlingmayer (1958) stated that the application of dieldrin pellets to a Florida salt marsh resulted in tarpon mortalities. Dieldrin has been identified from sediment samples from within the Tampa Bay estuary (Long et al. 1991); however, whether significant amounts of this or other toxic contaminants are present in tarpon tissues is not known.

3.1.4 ENVIRONMENTAL REQUIREMENTS

Data pertaining to the environmental requirements of tarpon are described in the following sections. Very little data were available for the Tampa Bay estuary specifically. Crabtree et al. (in press) report that directed sampling is necessary to obtain realistic estimates of tarpon distribution and abundance. Tarpon are rarely collected in generalized finfish surveys in the Tampa Bay estuary and therefore estuary-specific environmental requirements of this species have not been adequately addressed.

3.1.4.1 Turbidity

Early leptocephalus (Stage I) tarpon larvae occur only in clear offshore waters. Subsequent life history stages appear to be tolerant of high turbidities. Many habitats occupied by juveniles can generally be described as turbid and dark-stained (Zale and Merrifield 1989).

3.1.4.2 Water Temperature

Most collections of larval tarpon have occurred in offshore coastal waters where water temperatures are buffered from large temperature fluctuations. Surface water temperatures at larval collection sites in the Gulf of Mexico ranged from 26 to 30°C (Wade 1962; Berrien et al. 1978; Smith 1980; Cyr 1991; Crabtree et al. in press). Temperature at depth of capture were potentially as low as 22.2°C (Wade 1962).

Juvenile tarpon can tolerate a wide range of water temperatures. They have been collected in areas of relatively high water temperatures of 36-40°C (Moffett and Randall 1957; Rickards 1968; Wade 1969). Forty degrees appeared to be the upper temperature limit for juvenile tarpon (72-130 mm FL) acclimated at 25-27°C, and subjected to high laboratory test (gradually over 3 hrs) temperatures for 24 hours. All fish subjected to 39.4-39.6°C survived and those exposed to 40.5-41.9°C died (Moffett and Randall 1957). It is highly unlikely that water temperatures in Tampa Bay would meet or exceed these levels.

Juveniles are also collected in areas of cooler waters. Cyr (1991) collected juvenile tarpon near Naples, FL in water temperatures from 23 to 33°C, and in water temperatures of 17 to 37°C adjacent to Indian River. Wade (1969) collected juvenile tarpon in waters as low as 12°C.

The tarpon is a tropical and warm-temperate species that apparently cannot tolerate the large changes in temperature that can result from winter cold fronts. Juvenile tarpon recruitment and year-class strength may be linked to temperature-induced mortality. Shallow water in typical young-of-year habitat provides little buffer from air temperature fluctuations (Cyr 1991). Juvenile tarpon mortalities have occurred when air temperatures have dropped to -2.2°C at Sanibel Island, FL (Storey 1937; Storey and Gudger 1936) and

4°C at Jack Island, near Vero Beach, Florida (Crabtree, pers. comm. to Cyr 1991). Robins (1978) reported that the lower lethal temperature of tarpon is approximately 10°C. Tabb (pers. comm. to Wade 1962) observed dead tarpon at Everglades National Park when water temperatures dropped from 24°C to 11°C in several hours. Several tarpon died in holding tanks when water temperatures dropped from 21 to 12°C overnight (Rickards 1968). Episodic winter cold fronts are common in Florida, and the consequences of these events on tarpon year class strength could be significant (Cyr 1991).

3.1.4.3 Salinity

Egg and larval tarpon may require higher salinities than juveniles or adults. Early stage I (early leptocephalus stage) larvae have been collected only at oceanic salinities of 28.5-39.0 ppt (Wade 1962; Eldred and Lyons 1966; Eldred 1967,1968; Berrien et al. 1978; Smith 1980; cited from Zale and Merrifield 1989).

Juveniles are euryhaline and have been collected in areas where salinities ranged from 0 to 47 ppt (Hildebrand 1939; Simpson 1954; Moffett and Randall 1957; Harrington 1958; Harrington and Harrington 1961; Rickards 1968; Wade 1969; Dahlberg 1972;. Tagatz 1973; Tucker and Hodson 1976; cited from Zale and Merrifield 1989). Adults can be considered euryhaline and they are often collected in lagoons, ditches, canals, rivers, and estuaries with variable salinities (Wade 1962).

3.1.4.4 Dissolved Oxygen

Tarpon are highly tolerant of a wide range of dissolved oxygen concentrations. They are often found in stagnant land-locked pools with almost certainly low dissolved oxygen (Ellis 1956). These small shallow habitats occupied by tarpon may be connected to sources of water of higher water quality only intermittently at high tides (Wade 1962; Rickards 1968) or when impoundments are opened (Cyr 1991). Juveniles may occupy these areas of low dissolved oxygen and poor water quality as a method of avoiding predation (Wade 1962; Harrington 1966). Many fish predators would be unable to withstand these anoxic waters. However, these same water quality extremes may actually serve to limit juvenile tarpon growth (Cyr 1991). Extreme and variable dissolved oxygen and other water quality parameters may reduce growth efficiency (Doudoroff 1957; McKeown 1984; cited from Cyr 1991). Therefore, once the juvenile tarpon exceeds the high predation size threshold, they may find advantage in exiting the confining young-of-year habitat in search of better growth conditions (Cyr 1991). Schlaifer (1941) suggested that tarpon are obligate air breathers. In laboratory experiments, tarpon prevented from reaching the waters surface died from asphyxiation in 7-128 hours even in highly oxygenated water. The frequency of air breathing appeared to be inversely correlated with dissolved oxygen concentrations (Schlaifer 1941). However, more recent research conducted by S. Geiger at the University of South Florida has suggested that tarpon are not obligate air breathers and can survive at least two weeks without air breathing in well oxygenated water (R. Crabtree, pers. comm. 1992).

3.1.4.5 Water Depth

Leptocephalus tarpon larvae inhabit clear, warm, oceanic waters (Gehringer 1959; Robins 1978). Wade (1962) collected many in the upper 100 m of the water column, however sampling in that depth was more intense and may have biased estimates. Juvenile tarpon were collected in shallow (< 2 meters) estuarine waters (Wade 1962; Rickards 1968; Cyr 1991). Adults use a wide range of water depths, ranging from shallow waters to deep (90-1400 m) offshore spawning grounds (Cyr 1991). Most adults probably remain in the warmer surface waters of these deep areas because water temperature at deeper depths would be too low (R. Crabtree, pers. comm. 1992).

3.1.4.6 Structural Habitat

No data were available to describe critical structural habitats of tarpon in the Tampa Bay estuary. Juvenile tarpon habitat typically includes mangrove-marsh areas with soft muddy bottom sediments and no submerged vegetation. Cyr (1991) collected juvenile tarpon on the east (Jack Island, near Vero Beach) and west (near Naples) coasts of Florida in stagnant, murky pools with soft muddy unvegetated bottoms having a strong smell of hydrogen sulfide. These areas were surrounded primarily by mangroves. Similar habitats were described by many authors (Beebe 1927; Breder 1944; Wade 1962; Harrington 1966; Rickards 1968). Dominant habitat type at Sapelo Island, Georgia were sandy-mud unvegetated bottoms, with reeds along the shoreline.

Most young-of-year tarpon appeared to leave these habitats at approximately 400 mm SL, which corresponded to approximately one year of growth (Cyr (1991). This author speculated that this habitat shift may occur after tarpon reach a size threshold which allows them to avoid most predators. Increasing food requirements might also cause juvenile tarpon to leave these first-year habitats (Cyr 1991).

Development in south Florida has contributed to destruction and alteration of juvenile tarpon nursery habitat (Wade 1973). Mosquito control impoundments, particularly those along the Indian River, have reduced available habitat for juveniles by restricting access to these previously used areas. Only seasonally or permanently opened impoundments are available as nursery areas (Cyr 1991). Consequently, juveniles may be more susceptible to anthropogenic impact than other lifestages because they occur primarily within estuarine waters.

Very little is known about tarpon habitat preferences in the Tampa Bay estuary. The mangrove impoundment habitats described for the east coast do not exist extensively in this estuary. No juvenile or adult tarpon were collected in the fisheries-independent monitoring program conducted in Tampa Bay during 1989 and 1990 (FIMP 1989, 1990); however, types of sample gear used during this project are probably ineffective for collecting tarpon. Further studies, specifically directed at tarpon, are necessary to document critical structural habitat requirements of tarpon in the Tampa Bay estuary.

Environmental requirements of tarpon are summarized in Table **3-1.**

3.2 BAY ANCHOVY *(Anchoa mitchilli)*

3.2.1 INTRODUCTION

The range of the bay anchovy extends from Maine (Bigelow and Schroeder 1953) to Brazil (Hildebrand and Cable 1930) and along the Gulf of Mexico to Yucatan, Mexico (Hildebrand 1943). The bay anchovy is one of the most abundant fish species occurring in estuaries along the mid-Atlantic region and throughout the Gulf of Mexico (Wang and Kernehan 1979). Bay anchovy biomass may be higher than that of any other fish along the south Atlantic and Gulf coasts (Gunter and Hall 1963). As a widely distributed and abundant planktivore, bay anchovy represents a major link in estuarine food webs and probably plays a significant role both in survival patterns of benthic invertebrate larvae and in the dynamics of estuarine zooplankton community (Johnson et al. 1990). Because of their abundance and small size they are an important trophic link between plankton and piscivorous fish (Baird and Ulanowicz 1990). They are also important prey species for many commercial and recreationally important species (Robinette 1983; Vouglitois et al. 1987).

3.2.2 LIFE HISTORY

In the Tampa Bay estuary, bay anchovy spawning occurs from spring through fall months with peak egg densities occurring from April through July. Eggs were collected in the Little Manatee River area of Tampa Bay from March through November (Peebles, in prep.). During studies conducted in 1980 in Tampa Bay, spawning began after surface water temperatures reached 2O°C and ceased by November (Phillips 1981). Weiss and Phillips (1985) reported that egg and larval anchovies (primarily A. ***mitchi//i)*** were collected in ichthyoplankton surveys near Big Bend power station during March through October. Greatest egg densities occurred during April and May. Peebles et al. (1992) collected eggs and post-flexion bay anchovy larvae in Tampa Bay waters adjacent to the Little Manatee River mouth. Largest collections of juveniles and adults occurred upstream in the river. No bay anchovy eggs or larvae were reported in icthyoplankton collections at the mouth of Tampa Bay, although numerous striped anchovy ***(Anchoa hepsefus)*** were collected (Robison 1985). These studies suggest that the bay anchovy spawns within, rather than offshore, the Tampa bay estuary.

Table 3-1. General and preferred ranges and upper and lower tolerance limits for environmental requirements of tarpon. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	**Lower Limit**	**Upper Limit**	Range	Reference
Temperature				26-30°C (E, L)	Wade 1962; Berrien et al. 1978; Smith 1980; Cyr 1991; Crabtree et al. in press
(OC)				17-33 (J)	Cyr 1991
			39°C		Moffett and Randall 1957
		10 (J)			Robins 1978
		11 (J)			Tabb 1962
		12 (J)			Rickards 1968
Salinity (ppt)				28.5-39 (E, L)	Zale and Merrifield 1989
				0-47	Zale and Merrifield 1989
Dissolved Oxygen (mg/l)	tolerant of very low DO waters, potentially anoxic (J)				Wade 1962; Ellis 1956
Depth (m)	<2m (J)				Wade 1962; Rickards 1968; Cyr 1991
				in surface waters at depths to 1400 + (A)	**Cyr** 1991
				in surface waters at depths to 1400 + (A)	Cyr 1991
Substrate	soft mud, unvegetated bottoms, often mangrove areas (J)				Wade 1962, 1963; Harrington 1966; Rickards 1968, Cyr 1991

Larval, juvenile, and adult bay anchovy appear to be eurythermal and euryhaline. (Springer and Woodburn 1960; Robinette 1983). They are common throughout the year and at variable salinities in the Tampa Bay estuary (Haddad et al. 1992; Peebles et al. 1992). Juvenile and adult bay anchovy are commonly collected in shallow estuarine waters (<2 m); however, they have not been associated with particular substraits (Orth and Heck 1980; Gilmore 1987; Thayer et al. 1987).

Very little information was available to assess age and growth of bay anchovy in Tampa Bay estuary. In North Carolina estuaries, growth of larval bay anchovy was rapid; they grew from about 2.7 mm SL at hatching to 26.4 mm SL by the age of 49 days (Fives et al. 1986). In Tampa Bay, early juvenile stage begins at approximately 15 mm SL. Eggs have been reported in specimens as small as 20 mm SL (Peebles, pers. comm. 1992). Hellier (1962) examined growth of the bay anchovy in Laguna Madre, Texas. Fish averaging 25 mm were observed in March. They grew to 32 mm by August, 35 mm in September, and reached 45 mm by the following March. In North Carolina estuaries, the bay anchovy probably do not exceed 1.5-2 years of age (Fives et al. 1986). Springer and Woodburn (1960) constructed length-frequency tables for bay anchovy collected in Tampa Bay. Although very little information on growth rates was ascertained, there appeared to be two and sometimes three separate year classes. A major problem in examining bay anchovy growth is the protracted spawning season in Tampa Bay which makes distinctions between cohorts difficult. Peebles et al. (1992) reported early juvenile bay anchovy year round in the Little Manatee River (Fig. 3-2). Bay anchovy collected monthly at fixed stations throughout Tampa Bay during Florida's Fisheries Independent Monitoring Program (FIMP) also showed large overlap in average sizes (FIMP 1989, 1990).

No data were available to assess mortality rates of Tampa bay anchovy. In Biscayne Bay (FL), egg and larval bay anchovy mortality rates were high and variable ranging from 22 to 39 % per day (Leak and Houde 1987). A significant portion of the mortality experienced by the bay anchovy may occur during the egg stage; a period of 24 hours or less at temperatures greater than 26°C. Mean daily loss of larval bay anchovy cohort to predation (18-28%) was 2 to 3 times higher than losses to starvation (10-11%) (Leak and Houde 1987).

3.2.3 ECOLOGICAL ROLE

Numerous studies have documented feeding habits of the bay anchovy in various estuaries. The bay anchovy appears to be a selective feeder rather than a filter feeder and feeding is discriminatory as to both size and type of organisms (Detwyler and Houde 1970; Odum 1971; Johnson et al. 1990). In the North Inlet estuary in South Carolina, crab zoea (*Uca* spp.) and copepods comprised the majority of prey for adults (40-59 mm SL) (Johnson et al. 1990). In Lake Ponchatrain, (LA) juveniles (35-40 mm) fed primarily on microzooplankton (especially rotifers and copepods), detritus and organic matter. As they grew, they consumed greater amounts of macrozooplankton, especially mysids, small shrimp, and larval fishes, which collectively accounted for approximately 60% (by volume)

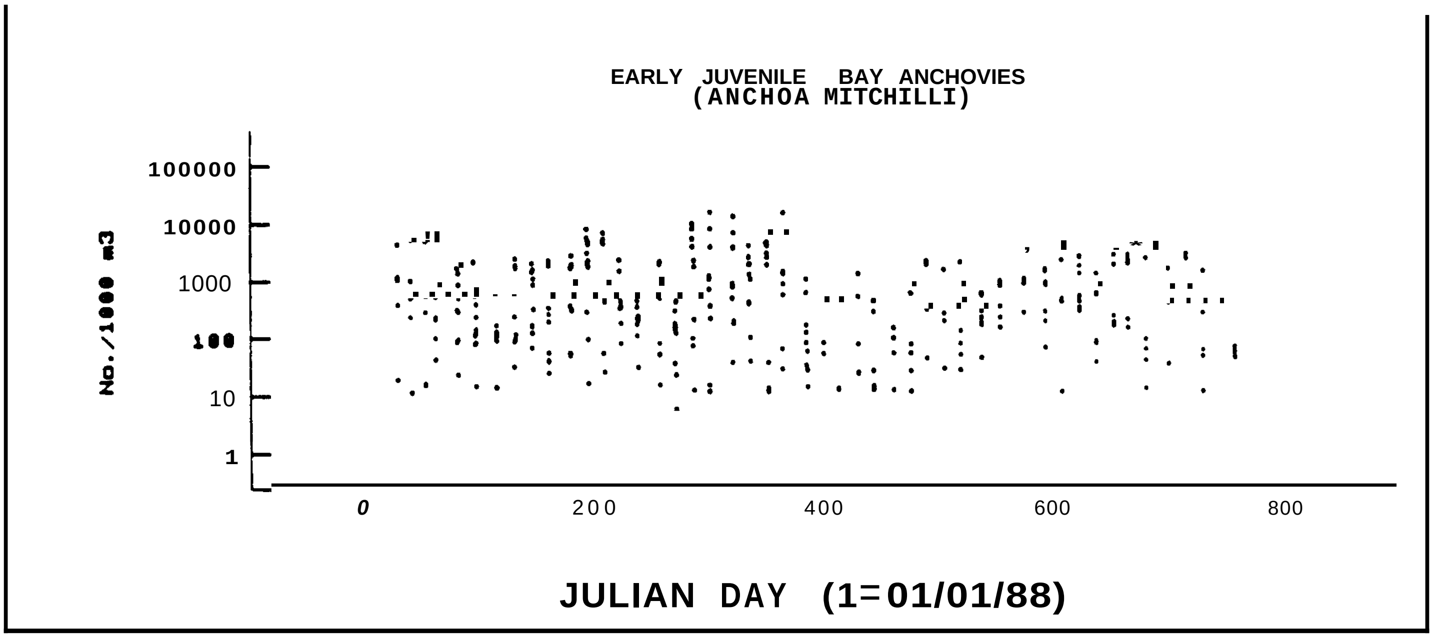

Figure 3-2. Seasonal abundance patterns observed for young bay anchovy in the Little Manatee River estuary. Source: Peebles et al. 1992.

of stomach contents of fish 60-65 mm long (Darnell 1958, 1961). In south Florida, Odum (1971) reported that bay anchovy less than 25 mm long primarily consumed planktonic copepods and copepod larvae. Fish 31-62 mm long fed on small benthic crustaceans, especially amphipods (22%), mysids (19%), harpacticoid copepods, ostracods and small mollusks, with lesser amounts of zooplankton and detritus. Near Crystal River, veligers (larval bivalves) accounted for 50-74% of the stomach contents of juvenile bay anchovy between 15-23 mm and large numbers of copepods were also consumed (Carr and Adams 1973). Springer and Woodburn (1960) reported that bay anchovy in Tampa Bay fed on copepods, ostracods, pelecypods, gastropods, crustacean larvae, mysids and unidentifiable fish. A thorough analysis of bay anchovy feeding habits in Tampa Bay suggests that calanoid copepods, gammarid amphipods, and decapod mysis larvae made up a large percentage (by volume) of food items in bay anchovy of approximately 16-50 mm SL. Copepod nauplii, diatoms, calanoid copepods, and invertebrate eggs made up a large percentage of number of organisms ingested (see Peebles 1992).

3.2.3.1 Predators

Their relatively small size and great abundance make the anchovies one of the most important groups of forage fish in the Gulf of Mexico (Robinette 1983). In Tampa Bay, numerous species are predators of the bay anchovy, including snook, spotted seatrout, white seatrout, gulf flounder, and lizard fish (Springer, and Woodburn 1960; McMichael et al. 1989; McMichael and Peters 1989; Peebles 1992).

3.2.4 ENVIRONMENTAL REQUIREMENTS

3.2.4.1 Salinity

Bay anchovies are euryhaline and have been collected in waters with salinities ranging from 0 to 45 ppt (Robinette 1983). The bay anchovy has a ubiquitous distribution in the Tampa Bay estuary and salinity appears to be of very little consequence in influencing distribution of juveniles and adults (Springer and Woodburn 1960). Juvenile and adult bay anchovy were collected throughout the Tampa Bay estuary in salinities ranging from 0.3 to 36.1 ppt as part of Florida's Fisheries Independent Monitoring Program during 1987 and 1990 (FIMP, unpubl. data 1992).

Bay anchovy spawning probably occurs in the higher salinity regions of the bay. Peebles et al. (1992) collected higher abundances of bay anchovy eggs and post-flexion larvae in Tampa Bay while juveniles and adults were more abundant in the Little Manatee River (Fig. 3-3). In the Delaware estuary, higher percentages of live eggs were found in higher salinity waters (20-30 ppt) than in low salinity waters (< 15 ppt) (Wang and Kernehan 1979).

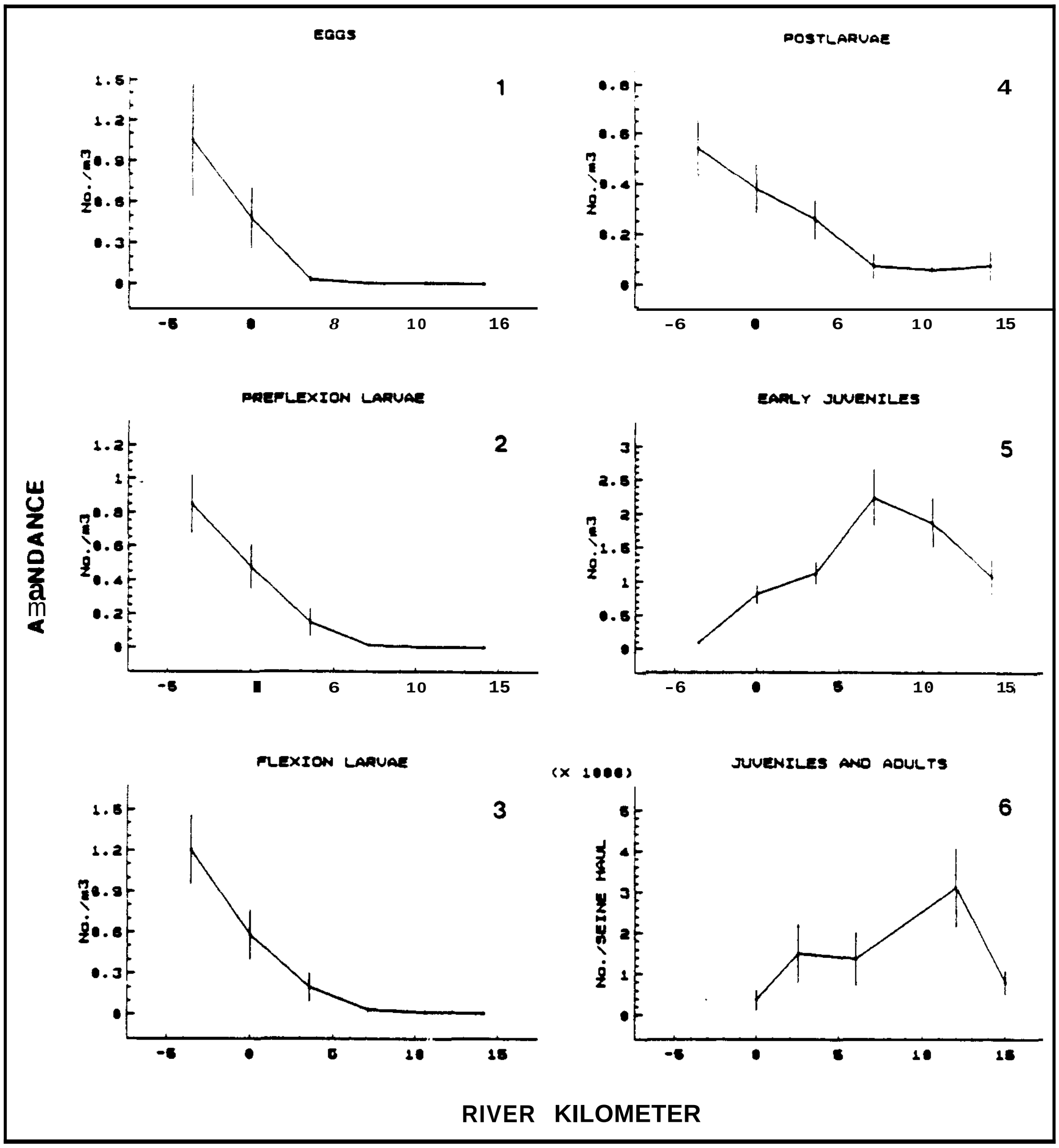

Figure **3-3.** Mean abundances and associated standard errors for anchovy developmental stages collected from the Little Manatee River. Age increases in the numerical order indicated. Preflexion and flexion stages are predominantly composed of bay anchovies ***(Anchoa mitchilli,*** but also contain striped anchovies ***(A. hepserus);*** all other stages are entirely composed of A. ***mitchilli.*** Eggs, larvae, and early juveniles were collected with a plankton net, juveniles and adults were collected with seines. Source: Peebles et al. 1992.

Bay anchovy distribution in the Little Manatee River area shows a pattern of decreasing salinity at capture with progressing larval and juvenile development (Peebles et al. 1992; Fig. 3-4). In an extensive juvenile and adult fish survey of the Little Manatee River, bay anchovies were commonly collected at all stations from the river mouth to freshwater tidal areas (Haddad et al. 1990; 1992). These data suggested a bimodality in anchovy distribution, with large numbers of individuals collected from the river mouth to rkm (river kilometer) 2.4 and again at rkm 11.8. This species was predominantly found in mesohaline (5-18 ppt) and oligohaline (0.5-5 ppt) waters within the river, but they were also common in polyhaline and freshwater areas (Fig. 3-5); Haddad et al. 1992).

Bay anchovies are found throughout the Tampa Bay estuary in a wide rage of salinities. Bay anchovy spawning and larval development may occur in the higher salinity ($>$ 18 ppt) regions of the Tampa Bay estuary. Juvenile and adult bay anchovy appear to be found throughout the estuary and tidal tributaries in freshwater to polyhaline salinities, with juveniles being particularly abundant in oligohaline and mesohaline waters.

3.2.4.2 Temperature

Bay anchovies in the Tampa Bay estuary are tolerant to a wide range of water temperatures. Springer and Woodburn (1960) collected bay anchovy in Tampa Bay at water temperatures ranging from 10.8-32.5°C. No studies have been conducted on the upper or lower tolerance limits of Tampa Bay anchovies, however, Chung (1977) reported upper temperature tolerance limits of bay anchovy from the Galveston, Texas area. Anchovies 20-70 mm had three-hour LD_{50}s in waters of 31-35°C, and 30-minute LD_{100}s in waters of 34-37°C. Galloway and Strawn (1975) observed that differences in relative abundance of bay anchovy in Galveston Bay may be due to temperature preferences. These authors suggested that this species preferred water temperatures of 24.5-32.5 °C and avoided temperatures of 33°C or higher. Bay anchovy collected in Galveston Bay during summer months (June-September) survived beyond three hours at 32°C, however, few fish lived more than three hours at 35°C and none survived for three hours at 36°C (Chung and Strawn 1982). These studies suggest that bay anchovy mortality may result from exposure to unusually high water temperatures, such as heated effluent from power plants.

Bay anchovies appear to be tolerant of average and below average water temperatures in the Tampa Bay estuary. No bay anchovies were reported in winter freeze kills in Tampa Bay (Rinckey and Salomen 1964; Gilmore et al. 1978). Bay anchovies remained in Tampa Bay throughout the year and often had peak abundances during winter months in the Little Manatee River (Haddad et al. 1992).

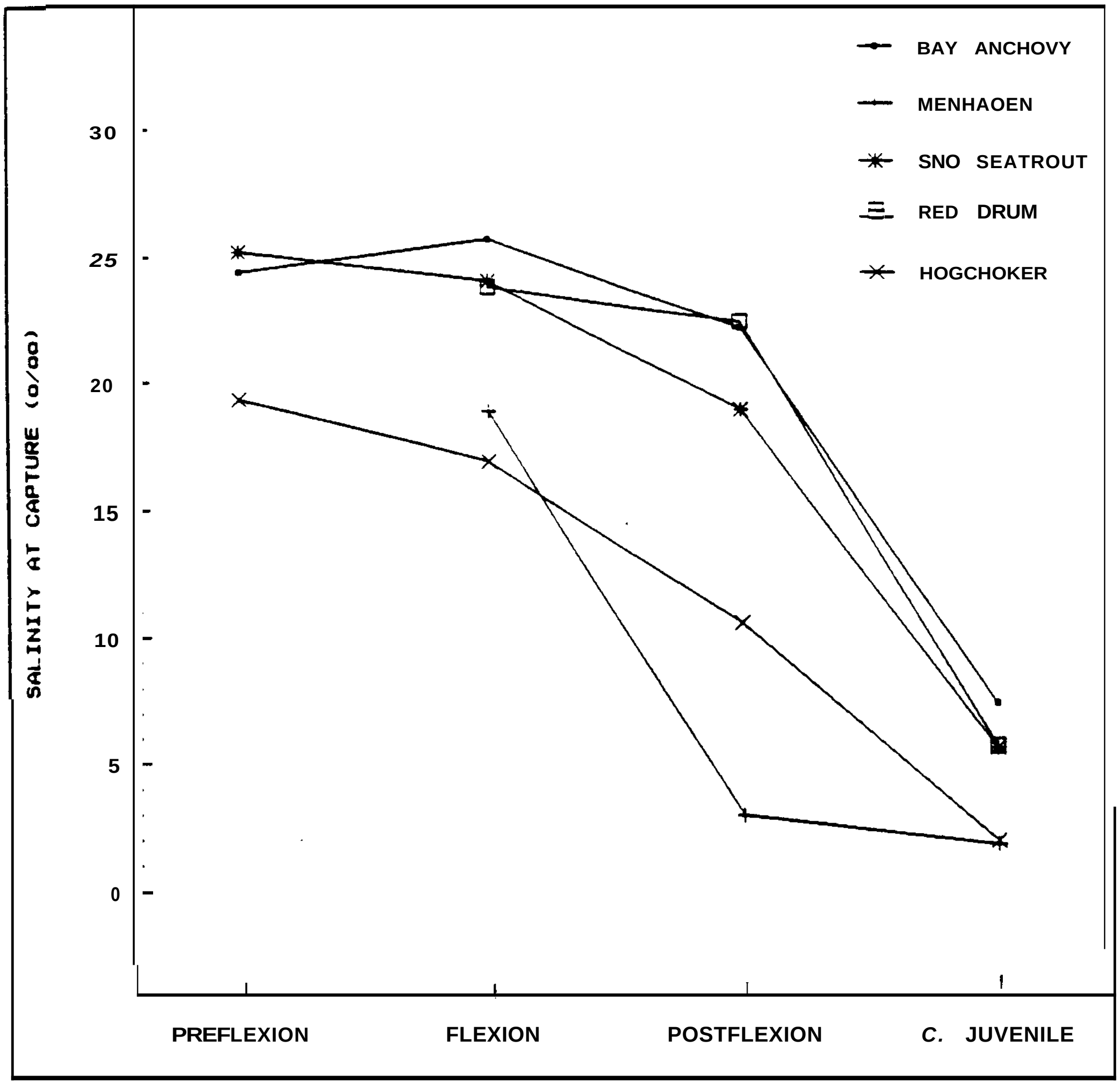

Figure 3-4. Examples of decrease in mean salinity at capture with progressing development. Source: Peebles et al. 1992.

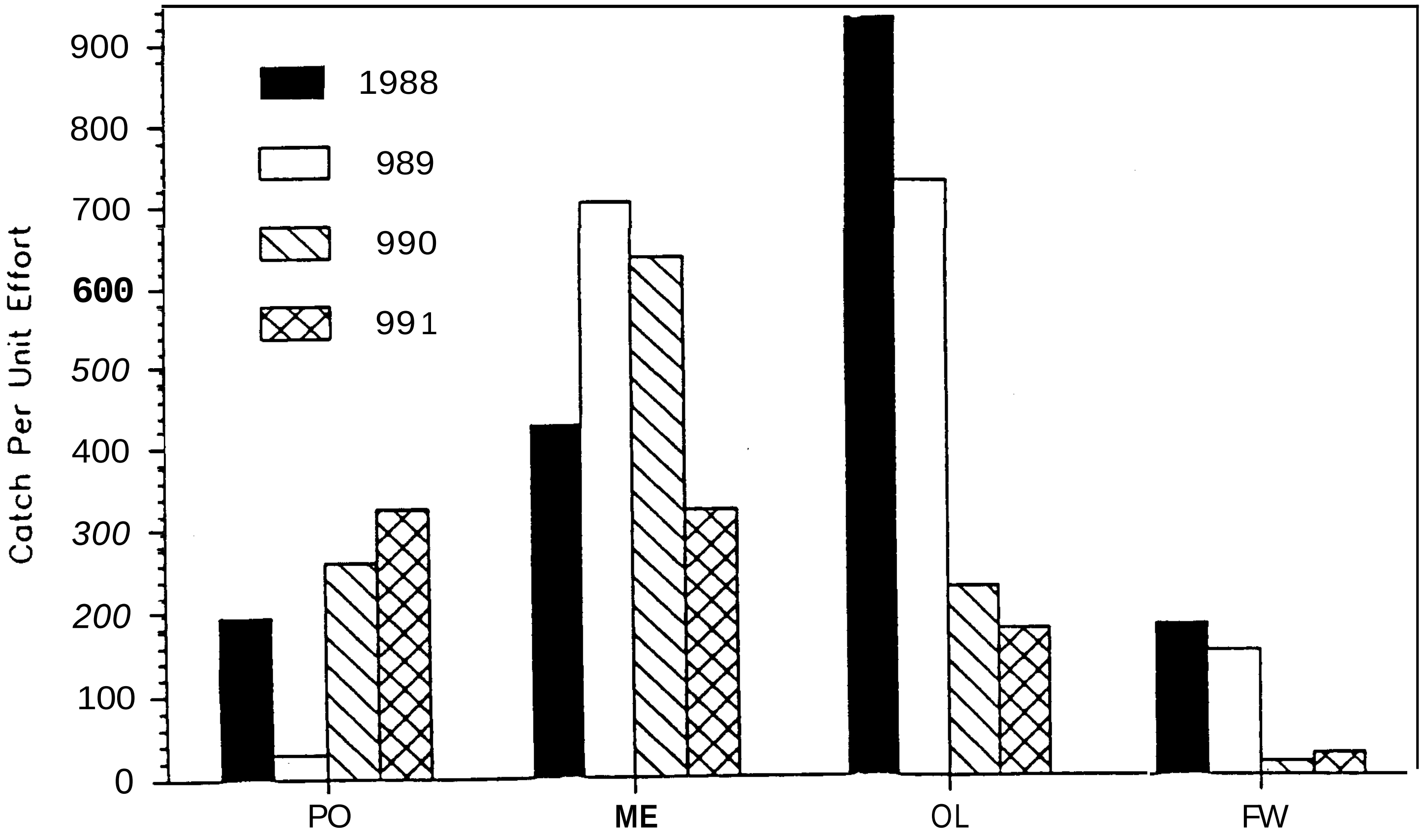

Figure 3-5. Juvenile and adult bay anchovy distribution in the Little Manatee River by salinity category. PO = polyhaline (> 18 ppt), ME = mesohaline (5-18 ppt), OL = oligohaline (0.5-5 ppt), and FW = freshwater (0-0.5 ppt). CPUE (number of fish/100 m^2). Source: Haddad et al. 1992.

3.2.4.3 Dissolved Oxygen

Very little information exists in which to document D.O. requirements of the bay anchovy in the Tampa Bay estuary. However, some information is available for the Chesapeake Bay area. D.O. concentrations below 3 mg L^{-1} probably limit the viability and productivity of bay anchovy in Chesapeake Bay. Laboratory experiments on bay anchovy eggs and yolk-sac larvae indicated that D.O. LC_{50} was 2.8 mg L^{-1} for eggs and 1.6 mg L^{-1} for yolk sac larvae (Chesney and Houde 1989). Egg hatchability declined significantly below 3.0 mg L^{-1} Survival of newly-hatched larvae declined below 2.5 mg L^{-1} Many 12-24 H posthatch larvae survived at concentrations between 2.0 and 2.5 mg L^{-1} and some survived when D.O. was between 1.0 and 2.0 mg L^{-1}, (Chesney and Houde 1989; cited from Houde and Zastrow 1991).

3.2.4.4 Water Depth

The bay anchovy is generally considered to have a ubiquitous distribution in shallow (< 2 m) waters (Gilmore 1987; Orth and Heck 1980). Bay anchovies are commonly collected in shallow (< 2m) waters in the Tampa Bay estuary (Springer and Woodburn 1960; Haddad et al. 1989, 1990, 1992; FIMP 1989, 1990).

Hildebrand (1963b) reported that the bay anchovy has been collected in waters as deep as 27-36 m, although it generally occurs in shallow depths. Little is known of the distribution of bay anchovy in deeper waters of the Tampa Bay estuary. In the Chesapeake and Delaware estuaries, juvenile and adult anchovy can occur throughout the water column (PSE&G 1984; Houde and Brandt 1990). Some evidence suggests that their preferred depth may be located nearer the surface than bottom (Kernehan et al. 1978). However, hydroacoustic surveys (Houde and Brandt 1990) indicate that changes in depth distribution occur, both seasonally and diurnally, that are not fully understood.

3.2.4.5 Structural Habitat

The bay anchovy has a widespread distribution in the shallow waters of many estuaries, however, they have not been associated with particular substrates or types of cover (Orth and Heck 1980; Gilmore 1987; Thayer et al. 1987). Bay anchovy were commonly collected in numerous habitats throughout Tampa Bay including those with vegetated and non-vegetated bottoms (Springer and Woodburn 1960; FIMP 1989, 1990).

Large numbers of bay anchovy were collected in seagrass beds ***(Halodule wrightii)*** at the mouth of the Little Manatee River. These fish showed no regular pattern in relation to seagrass density, which was expected, given the spatial and temporal patchiness of bay anchovy schools (Haddad et al. 1992). Bay anchovy and hogchoker ***(Trinectes maculatus)*** were the most abundant species in the oligohaline and freshwater reaches of the Little Manatee River (Haddad et al. 1990). However, water quality parameters are probably more important than structural habitats in determining their distribution within this system.

Bay anchovy collected at the mouth of the Little Manatee River showed some evidence of habitat preference, although the reason for this pattern was not fully understood. They were much less abundant at a station sheltered from the main river flow than at two stations directly in the rivers flow. Therefore, attraction to the river plume may influence their choice of habitats and affect distribution patterns (Haddad et al. 1992).

Environmental requirements of bay anchovy are shown in Table **3-2.**

3.3 STRIPED KILLIFISH *(Funduhs majalis)*

The killifish are comprised of three families: Cyprinodontidae, Anablepidae and Poecilidae (Rosen 1973). Two of these families, Cyprinodontidae and Poecilidae have species which are found in the Tampa Bay estuary (Springer and Woodburn 1960; Haddad et al. 1992; FIMP 1989, 1990; Table 3-3).

Relyea (1983) reports that the longnose killifish and the striped killifish, ***Fundulus majalis*** (commonly reported on the U.S. east coast) are the same species. The correct nomenclature for this species should be ***F. majalis*** (AFS 1991), however, many fishery reports in the Tampa Bay estuary still cite this species as F. ***similis*** (Matheson, per. comm. 1992). As a group, killifish are generally considered very tolerant of a wide range of environmental conditions. Martin and Finucane (1968) and Comp (1985) reported that the longnose killifish ***(Fundulus similis)*** is the most abundant cyprinodont in the Tampa Bay estuary.

The striped killifish ranges from New Hampshire south to Florida, and is found in the Gulf of Mexico (Rosen 1973; Hoese and Moore 1977). Although this species is not valued as a commercial or recreationally important fish, it is probably a very important component of the Tampa Bay ecosystem because of its abundance and distribution.

3.3.1 LIFE HISTORY

Striped killifish spawn in still, shallow water close to shore (Hardy 1978). During the breeding season, male striped killifish assume brighter colors (Ursin 1977). Spawning coloration and behavioral patterns are described in detail for this species in the Tampa Bay estuary (Springer and Woodburn 1960).

Spawning season appears to be extended in lower latitudes. In the Chesapeake Bay area, striped killifish spawn from April through September (Hardy 1978). Gunter (1945, 1950) reported ripe ***F. similis (=F. majalis)*** from March through August in Texas estuaries. In Tampa Bay, males in breeding colors were collected during February to

1

Table **3-2.** General and preferred ranges and upper and lower tolerance limits for environmental requirements of bay anchovy. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	**Lower Limit**	**Upper Limit**	**Range**	**Reference**
Temperature (°C)	**>20** (E,L)				Phillips **1981**
				10.8-32.5 (J,A)	Springer and Woodburn **1960**
	24.5-32.5 (J,A)		**33** (avoidance)		Galloway and Strawn **1975**
			35-36		Chung and Strawn **1982**
Salinity (ppt)				**0-45**	Robinette **1983**
				0.3-36.1 (J,A)	FIMP unpubl. data
	>**18** (L)				Peebles et **al. 1992**
	0.5-18 (J,A)				Haddad et al. **1990, 1992**
Dissolved Oxygen (mg/l)				**1.0-15.2** (J,A)	FIMP unpubl. data
		3.0 (E,L)			Chesney and Houde **1989**
Depth **(m)**	**<2** m (J,A)				Springer and Woodburn **1960** Orth and Heck **1980;** Gilmore **1987;** Haddad et al. **1989, 1990, 1992;** FIMP **1989, 1990**
Substrate	None				Springer and Woodburn **1960** Orth and Heck **1980;** Gilmore **1987;** Haddad et al. **1992**

Table 3-3. List of killifish species collected from the Tampa Bay estuary

Adenia xenica	Diamond killifish
Cyprinodon variagatus	Sheepshead minnow
Floridichthys carpio	Goldspotted killifish
Fundulus grandis	Gulf killifish
Fundulus majalis	Striped killifish
Gambusia affinis	Mosquitofish
Lucania parva	Rainwater killifish
Poecilia latipinna	Sailfin molly
Fundulus seminolis	Seminole killifish
Lucania goodei	Bluefin killifish
Fundulus chrysotus	Golden topminnow
Jordanella floridae	Flagfish
Heterandria formosa	Least killifish

September (Joseph and Yerger 1956; Springer and Woodburn 1960). Martin and Finucane (1968) suggested that spawning probably occurs year round in the Tampa Bay estuary, although it appeared to peak in spring and late fall and was reduced from July through September. Courtship, spawning, and embryonic development have been described from laboratory observation of fish collected in the Tampa Bay estuary (Martin and Finucane 1968). Ripe females in the mid-Atlantic region contained approximately 460-800 eggs suggesting that this species has a fairly low fecundity (Hardy 1978).

Very little information was available on larval striped killifish. Juveniles and adults had a ubiquitous distribution throughout the Tampa Bay estuary; however, they appeared to prefer shallow, higher salinity waters along the shoreline of the Bay (Springer and Woodburn 1960; Martin and Finucane 1968; Haddad et al. 1992; Peebles et al. 1992; FIMP unpublished data).

Information on growth of *F. majalis* is limited. Springer and Woodburn (1960) observed a definite bimodality at lengths of 27 and 53 mm for three months consecutively (August -October) and evidences of them were also present in earlier months. However, peaks at lesser or greater sizes appeared and disappeared randomly. Martin and Finucane (1968) reported that growth rate of *F. similis* was rapid, but because of the protracted' spawning period, overlapping size classes, and possible sex differences; growth curves were not determined. Seasonal length-frequency distribution of *F. similis* collected in Tampa Bay are shown in Table 3-4.

Size at maturity for *F. majalis* was reported at 76 mm and 64 mm for females and males, respectively (Hildebrand and Schroeder 1928). This species appeared to reach maturity by at least their second year.

3.3.2 ECOLOGICAL ROLE

Fundulus majalis (reported as *F. similis)* feed primarily on copepods, ostracods, tiny molluscs, and annelids in Tampa Bay (Springer and Woodburn 1960). In the Chesapeake Bay, this species feeds primarily on small crustaceans and polychaetes, although they appeared to use all available food items except detritus (Baker-Dittus 1978). This author reported that differences in feeding ecology and seasonal distribution of three sympatric species of *Fundulus,* including *F. majalis,* functioned in reducing the potential interaction among the species.

3.3.2.1 Predators

Small estuarine fishes such as killifish are major prey for many bird and fish species (Peterson and Peterson 1979). Birds which feed on killifish include herons, egrets, terns and gulls (Valiela et al. 1977). Striped killifish are probably also prey of larger predatory fish species. Schomer and Johnson (1990) report that snook, tarpon, ladyfish, and other piscivorous species feed on killifish in the Tampa Bay estuary.

Table 3-4. Frequency distribution of *F. similis* by standard lengths in Tampa Bay, 1962

Midclass (mm)	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec	
15	1			9	4	1				3	7		
20	1	1	1	49	19	28				27	56	8	
25	1		4	83	28	54	5		4	64	67	22	
30	9	2	4	45	47	55	75	8	6	51	102	25	
35	4	2	14	14	112	34	234	49	16	45	100	40	
40	7	7	31	16	126	19	198	70	58	27	106	58	
45	3	8	61	9	35	8	100	93	76	32	84	71	
50	2	10	31	9	23	2	47	75	73	49	44	36	
55	3	6	21	20	22	6	30	40	47	47	22	49	
60	4	5	16	5	11	8	9	26	30	44	23	43	
65	10	7	14	5	11	6	8	20	28	37	17	28	
70	10	10	18	12	15	6	15	17	16	27	12	20	
75	9	6	13	1	7	7	14	11	5	14	10	20	
80	8	8	19	2	11	6	22	15	2	11	7	13	
85	4	6	6	1	12	4	15	13	2	26	1	5	
90	11	2	2	2	8	5	12	4	3	18		1	
95	3	1	3		2	4	12	6	2	12		1	
100	2		1		5	3	16	2		9			
105	2				2	3	9		3	9		2	
110						1	1						
115									1				
Total	94	81	259	272	500	260	822	450	372	552	658	442	4762
Average standard length (mm)	64.7	61.1	54.5	33.2	43.8	40.4	48.2	51.6	51.5	57.9	40.0	49.4	

Source Martin and Finucane (1968)

3.3.3 CONTAMINANTS

No information was available to document contaminant concentrations in striped killifish.

3.3.4 ENVIRONMENTAL REQUIREMENTS

Killifish are typically very tolerant of extremes in environmental variables such as salinity, temperature and dissolved oxygen. Although this species has a ubiquitous distribution throughout estuary, it appears to have a preference for shallow, higher salinity waters along the shoreline portion of the bay. Specific environmental requirements will be discussed in detail in the following sections.

3.3.4.1 Salinity

Many studies have suggested that this species is euryhaline, although it prefers more saline portions of estuaries (Martin and Finucane 1968; Rosen 1973; Robins et al. 1980). Simpson and Gunter (1956) reported that these species were common in salinities up to 76.1 ppt. In Tampa Bay, *F. majalis* (reported as *F. similis)* were collected in salinities ranging from 0.3 to 35 ppt, although most were collected in main bay region at salinities greater than 25 ppt (FIMP, unpubl. data). *Fundulus majalis* exhibited a very consistent distributional pattern within and among years in the Little Manatee River region (Haddad et al. 1992). Within each year, this species was always most abundant at the mouth of the river and at polyhaline salinities (Figs. 3-6 and 3-7). A secondary area of abundance occurred from rkm 2.5 to 5.8 in more mesohaline salinities (5-18 ppt). They were collected in oligohaline and freshwater regions of the river, although they were less abundant than at higher salinities. This consistent pattern in spatial distribution of *F. majalis* within the Tampa Bay system suggests that salinity may play a major role in influencing the distribution of this species within the estuary.

3.3.4.2 Temperature

Like many species of killifish, F. majalis is eurythermal. This species does not appear to be susceptible to extreme cold water conditions which have caused mortalities in other fish species. Healthy *F. similis (=F. majalis)* were collected in Old Tampa Bay and at Pinellas Point during December 1962 in water temperatures of 9.6°C, and 11.2°C, respectively (Rinckey and Salomen 1964). However, this species is less common in shallow coastal regions during the winter months, suggesting an avoidance of cooler water temperatures. Springer and Woodburn (1960) reported that this species was more abundant from late spring to early fall and they collected no specimens from February through April. In the Little Manatee River region, *F. majalis* was always more abundant in summer months than during fall or winter (Haddad et al. 1992, Fig. 3-8). Similar distribution patterns were noted in McKay Bay (Price and Schlueter 1985). These authors mentioned that low numbers in winter collections may have been the result of movement from colder, shallow waters to more temperature-stable deeper channels.

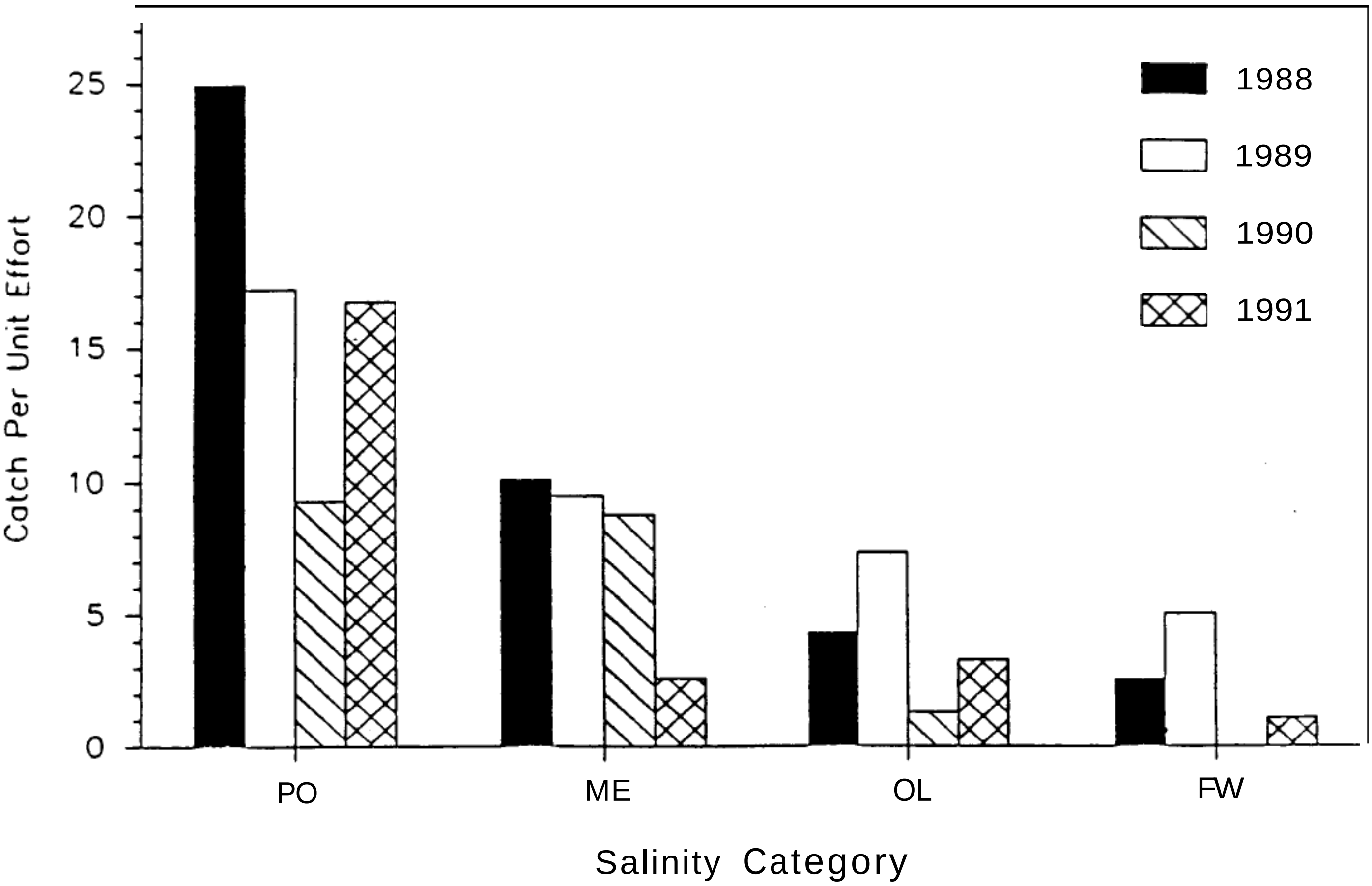

Figure 3-6. CPUE (number of fish/100 m^2) for juvenile and adult *F. majalis* by surface salinity in the Little Manatee River. PO = polyhaline (> 18 ppt), ME = mesohaline (5-18 ppt), OL = oligohaline (0.5 - 5 ppt), and FW = freshwater (0 - 0.5 ppt). Source: Haddad et al. 1992.

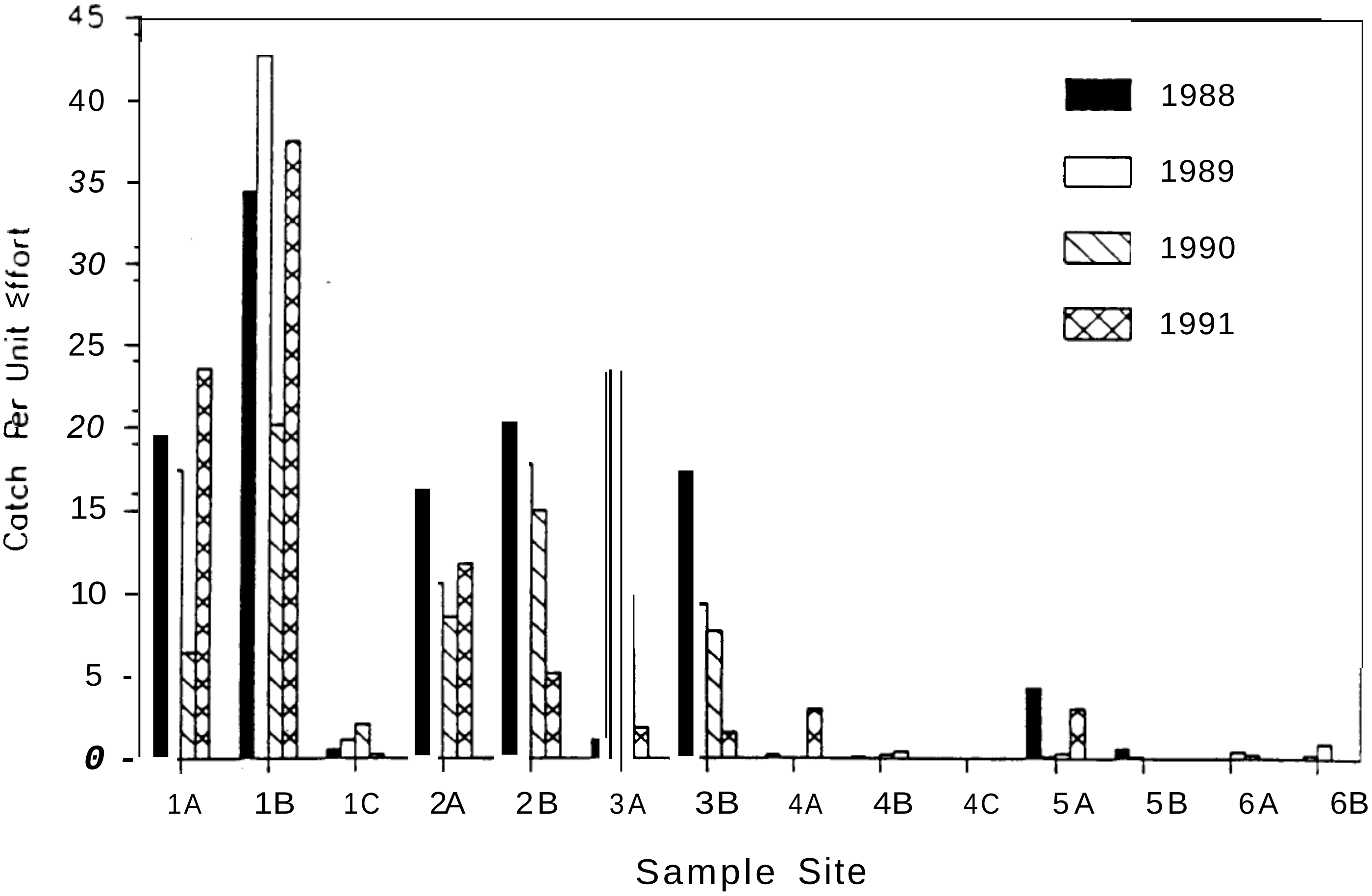

Figure 3-7. **CPUE (number of fish/100 m^2) for juvenile and adult *F. majalis* by station location in the Little Manatee River. Station 1 = rkm 0, station 2 = rkm 2.5, station 3 = rkm 5.8, station 4 = rkm 11.8, station 5 = rkm 15.5, and station 6 = rkrn 16.4. Source: Haddad et al. 1992.**

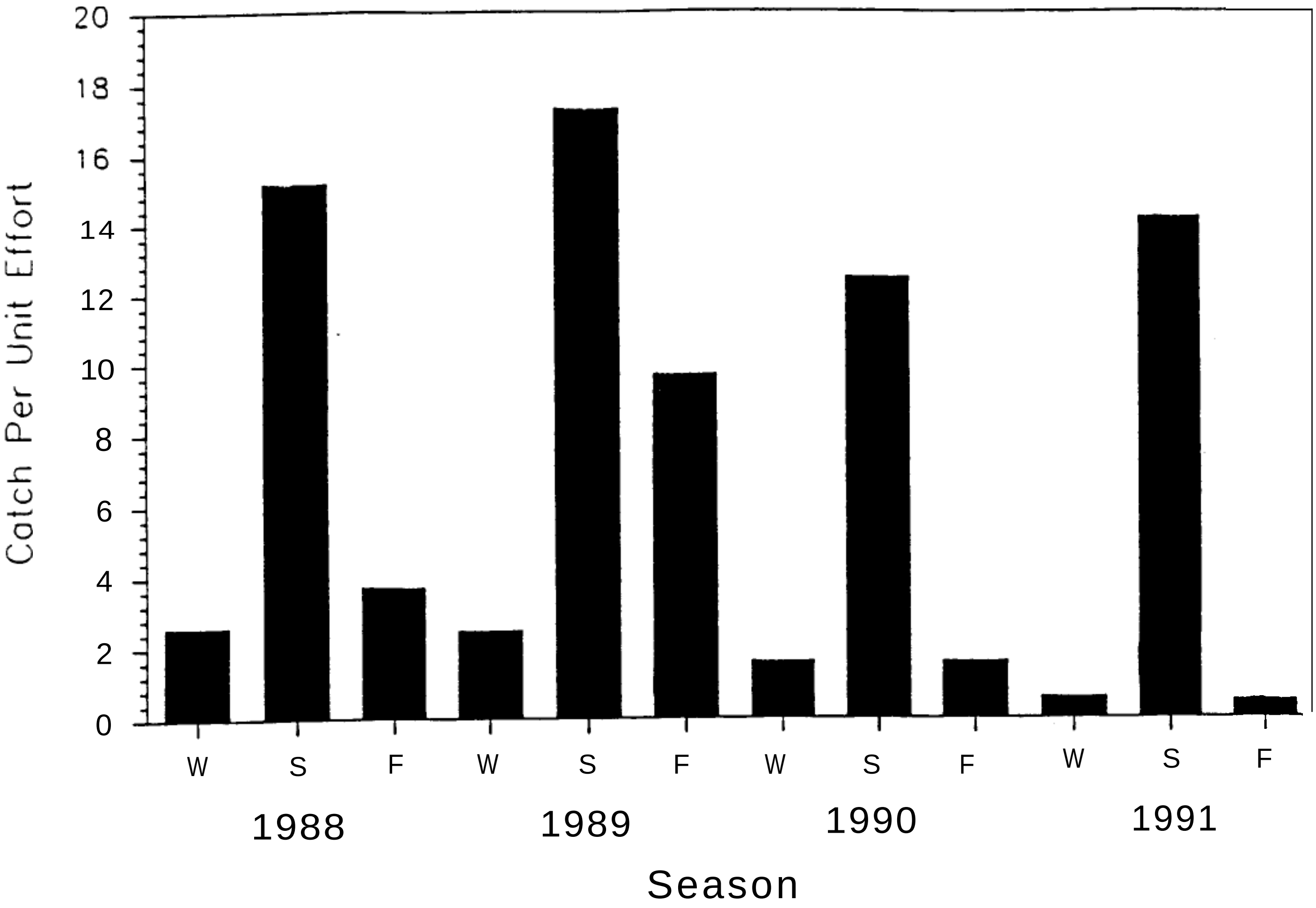

Figure 3-8. **Seasonal abundance of juvenile and adult *F. majalis* by season in the Little Manatee River. Source: Haddad et al. 1992.**

More data are needed to assess upper temperature limits of this species in the Tampa Bay estuary; however, these limits have been examined in laboratory studies. Striped killifish mortality was reported in 63 minutes at 34°C, 28 minutes at 36°C, 9 minutes at 37°C and 2 minutes at 42°C (Orr 1955; cited from Abraham 1985). Martin and Finucane (1968) noted a decline in abundance of small juveniles during June-September (Table 3-4) and stated that water temperatures greater than 30°C may depress spawning. These authors collected this species in Tampa Bay in water temperatures as high as 33.3°C. These studies suggest that striped killifish appear to be tolerant of average water temperature extremes present in the Tampa Bay estuary; however, they do show patterns of reduced abundance in cooler, shallow waters during winter months, suggesting that cold water temperatures, although not lethal, may influence their distribution within the estuary.

3.3.4.3 Dissolved Oxygen

No data were available to document dissolved oxygen tolerances of this species. Juvenile and adult *F. similis* have been collected in Tampa Bay waters with dissolved oxygen concentrations ranging from 1.4 to 11.1 ppm (FIMP, unpubl. data), which suggests that they may be tolerant of low D.O. conditions. Additional studies are needed to document lethal limits.

3.3.4.4 Water Depth

In Tampa Bay, juvenile and adult *F. majalis* prefer shallow estuarine waters usually along the land/water interface (Springer and Woodburn 1960). Data collected as part of the Florida Fisheries Independent Monitoring program shows a predominance of *F. similis* (=*F, majalis)* in seine collections along shallow (< 0.5 m deep) shorelines throughout the Tampa Bay estuary. *Fundulus similis* was often the second or third most commonly collected species in these collections. They were collected less frequently with offshore seines, and almost never with trawls, both of which sampled deeper waters (FIMP 1989, 1990). The large proportion of *F. similis* collected adjacent to the shoreline relative to other shallow (0.5 -2 m) areas emphasizes their preference for these very shallow regions.

3.3.4.5 Structural Habitat

Striped killifish abundance and distribution is apparently not influenced by submerged aquatic vegetation (SAV). *Fundulus rnajalis* are commonly collected over both vegetated and non-vegetated bottoms (Haddad et al. 1992; Price and Schlueter 1985; FIMP 1989, 1990). Springer and Woodburn (1960) report that they appear to prefer sandy or sandy mud bottoms within Tampa Bay. Martin and Finucane (1968) found that both juvenile and adult longnose killifish (= striped killifish) preferred soft sandy-silt bottom and an open beach usually free of attached seagrasses. Bottom composition appeared important since soft sediment allowed the fish to burrow to escape predation.

Fundulus similis was commonly collected in McKay Bay over unvegetated bottoms comprised of fine sand and silt (Price and Schlueter 1985). The McKay Bay site was lined with stands of black mangrove, *Avicennia germinans,* white mangrove, *Laguncularia racemosa,* patches of smooth cordgrass, *Spartina alterniflora,* as well as open sand mud beach areas, suggesting that shoreline vegetation probably has little influence on their distributions.

Environmental requirements of striped killifish are summarized in Table 3-5.

3.4 SNOOK *(Centropomus undecimalis)*

3.4.1 INTRODUCTION

Snook are distributed in the coastal waters of the tropical and subtropical western Atlantic Ocean, from North Carolina to Brazil, including the Gulf of Mexico and Caribbean Sea. In the Gulf of Mexico, they occur south of Tampa, Florida and Galveston, Texas, but rarely in the northern Gulf (Baugman 1945; Marshall 1958; Hoese and Moore 1977).

Four species of *Centropomus* occur in Florida waters (Rivas 1962, 1986). *Centropomus undecimalis* is the most common and ubiquitous of the four (Rivas 1962; Seaman and Collins 1983). Snook are an important recreational species in Florida, particularly along the lower Gulf coast. A relatively short-lived commercial snook fishery existed in Florida in the mid-twentieth century. Market demand for this species increased after 1930 and peaked in response to World War II food shortages (Marshall 1958). In the mid-1950s perceived declines in the snook populations were attributed to commercial fishing and in 1957, the State of Florida eliminated the commercial snook harvest (Marshall 1958; Seaman and Collins 1983). Declining snook populations were attributed to increased fishing pressure and deterioration of habitat (Hoese and Moore 1977; Seaman and Collins 1983). Marshall (1958) believed that reductions in snook populations could not be blamed entirely on fishing mortality. He believed that environmental alterations and degradations including reduced freshwater discharge into estuaries, sewage and industrial pollution, dredging and filling, mosquito control practices and insecticide use also contributed to population declines.

3.4.2 LIFE HISTORY

The snook is essentially a non-migratory species although they do undergo short movements to spawning areas. Approximately 90% of snook recaptured during tagging studies near the Naples/Marco Island area of Florida were caught less than 10 miles from their tagging site (Bruger, pers. comm. 1992). Snook spawning occurs near the mouths of rivers, canals, and passes and along adjacent coastal shorelines (Marshall 1958). Volpe (1959) collected ripe snook just offshore of the sandy beaches in the mouths of various saline open water passes along south Florida's Gulf coast during June and July. Probable snook spawning in the Tampa Bay estuary occurs at major inlets and at mouths of major embayments in the southern portion of Tampa Bay. These areas include: Rattlesnake Key at the mouth of Terra Ceia Bay, Port Manatee Spoil Island, Mari Posa Key at the mouth of Bishops Harbor and Mermaid Point at the mouth of Riviera Bay (Taylor et al., in prep.).

Table 3-5. General and preferred ranges and upper and lower tolerance limits for environmental requirements of striped killifish. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature **(°C)**		<9.6°C (J,A)			Rinckey and Salomen 1964
				to 33°C (J,A)	Martin and Finucane 1968
			34°C		Orr 1955
Salinity (ppt)				to 76	Simpson and Gunter 1956
				0-35 (J,A)	FIMP unpubl. data
	>25 (J,A)				FIMP unpubl. data
	>18 (J,A)				Haddad et al. 1992
Dissolved Oxygen (mg/L)				1.4-11.1 (J,A)	FIMP unpubl. data
Depth (m)	<0.5 (J,A)				FIMP 1989, 1990
Substrate	sandy-mud (J,A)				Springer and Woodburn 1960
	sandy-silt (J,A)				Martin and Finucane 1968
				unvegetated to vegetated (J,A)	Haddad et al. 1992; FIMP 1989, 1990

In Tampa Bay, Gonadosomatic indices (GSI) suggested snook spawning occurred from May to September and April to September during 1988 and 1989, respectively (Taylor, pers. comm. 1992). Fore and Schmidt (1973) reported that spawning occurred during May to mid-November in the Ten Thousand Islands area along the southwest coast of Florida. This suggested that a more protracted spawning season may occur in more tropical waters.

Many studies have attempted to collect larval snook in south Florida, although until recently, only a few were captured (Gilmore et al. 1983; Tolley et al. 1987; Rutherford et al. 1986; McMichael et al. 1989). Peters et al. (1992) collected 40 late stage snook larvae in the Little Manatee River during 1990 and 1991. Dozens of larvae were also observed among the red mangrove prop roots in lower Tampa Bay. Recent investigations in Tampa Bay have suggested that large numbers of larval and early juvenile snook use marginal habitats of the Tampa Bay estuary as nursery areas (Edwards 1990; Haddad et al. 1992; Peters et al. 1992). Salinity does not appear to be a significant factor influencing larval and juvenile snook distribution. However, factors such as water temperature, water depth, currents, and availability of structural habitat probably play an important role in choice of nursery habitats (Haddad et al. 1992; Peters et al. 1992). Typical nursery and adult habitat includes mangrove and backwater areas, tidal tributaries and deeper channels (Taylor, pers. comm. 1992).

Age and growth parameters have been described for juvenile and adult snook in Tampa bay. Growth rates of juvenile snook in Tampa Bay were approximately 0.6 to 0.7 mm per day, with highest growth rate at 1.2 mm per day (McMichael et al. 1989). These growth rates are similar but less than that reported for other areas in Florida. Fore and Schmidt (1973) reported that juvenile snook grew at 0.9 mm per day in the Ten Thousand Island area. Juvenile snook in Indian River grew at approximately 1.0 mm per day (Gilmore et al. 1983). Volpe (1959) reported that growth rates of snook in southwest Florida are relatively high to the second year, slower and more uniform to the fifth year and then reached an asymptote. Maximum ages determined for Tampa Bay snook were 19 and 13 years, for females and males, respectively (Taylor, pers. comm. 1992). Von Bertalanffy growth curves show significant differences in growth between the sexes. Asymptotic sizes were 1242 mm FL and 689 mm FL for females and males, respectively (Taylor, pers. comm. 1992).

Recent research suggests that Tampa Bay snook undergo a sex reversal with growth. They appear to be protandric hermaphrodites, changing from male to female (Taylor and Grier 1991). Some fish examined had both male and female gonadal material. Of 1,870 adult snook collected by these authors in Tampa Bay during 1988 and 1989, male lengths were 175 to 925 mm FL and females were 425 to 1100 mm FL.. The ratio of males to females for snook less than 500 mm FL was 5.5M:IF, for fish greater than 800 mm FL, it was 6.3F: 1.0M. Based on gonad histology, no females were less than 400 mm FL, nor were any age 0 or age 1 females observed (Taylor, pers. comm. 1992). Similar sex reversals may occur in snook populations from other areas. Similar differences in sex ratios relative to snook age were observed in Everglades National Park (ENP). The snook sex ratio in ENP favored males 11:1 at age 2 to 2:1 females at age 8. This difference in sex ratios had been attributed to sexual differences in annual mortality rates (Thue et al. 1982).

Size at maturity of Tampa Bay snook varied between the sexes. Fifty percent of males were mature by 401 mm FL. Fifty percent of females were mature by 499 mm FL. One hundred percent of males were mature by 575 mm and 100% of females matured by 750 mm. Fifty percent of males and females were mature by age 2+ and age 3+, respectively. One hundred percent of males and females were mature **by** age 5 + and age 7 +, respectively (Taylor, pers. comm. 1992).

3.4.3 ECOLOGICAL ROLE

Snook are opportunistic carnivores, they tend to be piscivorous, and specific diets are dependent upon the habitat in which they reside (Seaman and Collins 1983). Juvenile snook collected in Tampa Bay showed evidence of ontogenic changes in diet at sizes of approximately 45 mm SL (McMichael et al. 1989). Juveniles less than 45 mm obtained most of their prey volume from mysids (primarily *Mysidopsis almyra)* and copepods (primarily *Acartia tonsa).* Juveniles greater than 45 mm SL consumed some mysids but obtained most of their prey volume from fish (Cyprinodontids and Poecillids) and shrimp (Palaemonids) (Fig. 3-9). These findings are consistent with other juvenile snook (25 to 120 mm) feeding ecology studies along south Florida coasts which suggest they primarily consume fish, palaemonid shrimp and macrocrustaceans (Harrington and Harrington 1961; Fore and Schmidt 1973; Gilmore et al. 1983).

Adult snook feeding ecology is currently being investigated in Tampa Bay (Taylor, pers. comm. 1992). Adult snook in the Ten Thousand Islands area of Florida fed primarily on fish and crustaceans,(Fore and Schmidt 1973). Fishes represented approximately 50% of the adult diet (by occurrence and volume), with the majority comprised of pinfish *(Lagodon rhomboides)* **,** scaled sardine *(Harengula jaguanal) ,*mojarras *(Eucinostomus* spp.), lizardfish *(Synodus foetens),* and gold spotted top minnow *(Floridichthys carpio)* **.** Crabs and shrimp were of greater importance in the diet of adult snook than juveniles. Marshall (1958) reported that the wide range of prey of adult snook is attributable to the wide range of salinities in which snook are found.

3.4.3.1 Competitors

Studies documenting competitors and predators of snook are limited. Fore and Schmidt **(1**973) suggested that redfin needlefish *(Stronglyura notata),* Florida gar *(Lepisosteus platyrhincus)* and other young-of-the-year snook were of sufficient size to prey upon juvenile snook. Predators of snook in Tampa Bay include other snook, barracuda, and bottlenose dolphins (Taylor, pers. comm. 1992).

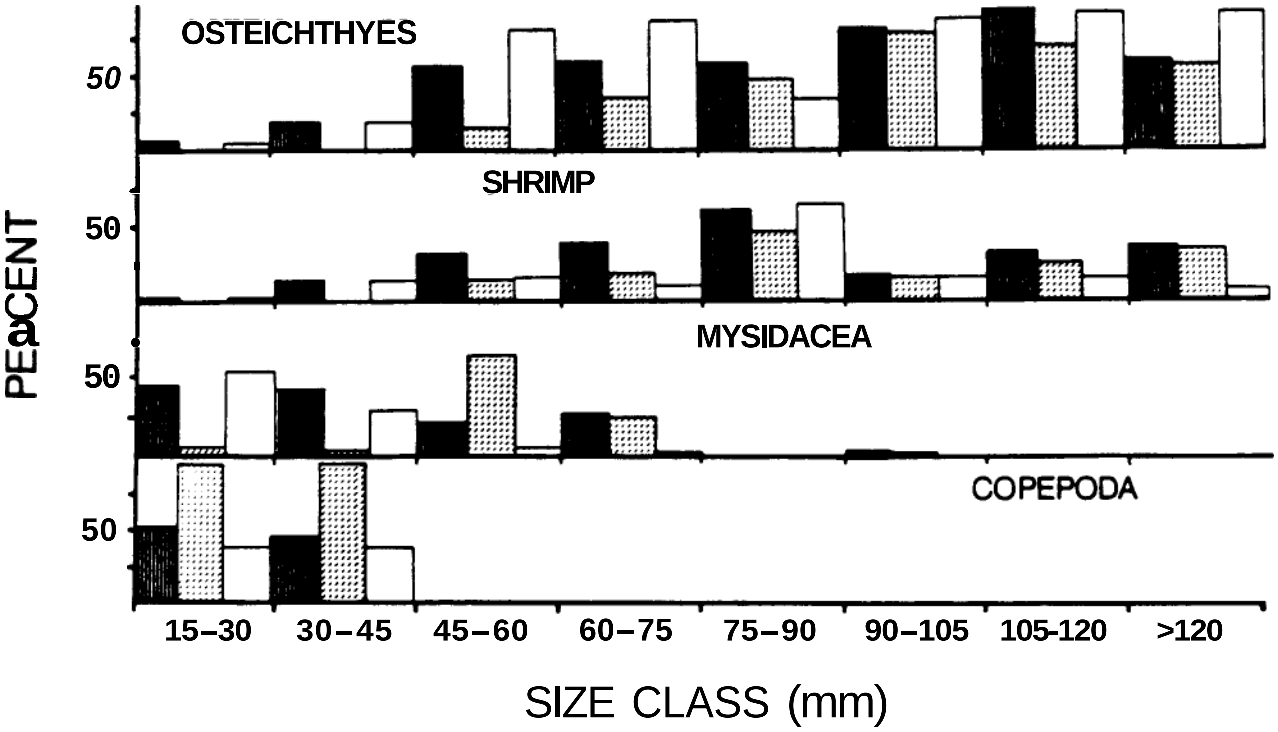

Figure 3-9. Percentages of volume (shaded), number (stippled), and occurrence (open) of major prey groups for each 15-mm-SL size class of snook collected in Tampa Bay. Source: McMichael et al. (1989).

3.4.4 CONTAMINANTS

Only limited information is available on contaminants in snook. Concentrations of four chlorinated hydrocarbons (DDE, DDT, DDD and PCBs) were found in two subadult and 1 adult snook collected from the Ten Thousand Islands area (Fore and Schmidt 1973). No detectable concentrations of 19 other pesticides were found, including aldrin, chlordane, deldrin, and mirex. Both DDT and PCBs were detected in sediment samples from the Tampa Bay estuary (Long et al. 1991). As an apex predator in the Tampa Bay estuary, it is possible that these chlorinated hydrocarbons could become concentrated in snook tissues. No data are currently available to evaluate this situation; however, ongoing fish tissue analyses being conducted by FDNR-FMRI in the Tampa Bay estuary may provide some of this information (McMichael, pers. comm. 1992).

3.4.5 ENVIRONMENTAL REQUIREMENTS

The Tampa Bay estuary provides many of the water quality and structural habitats required by snook throughout their life history. Salinity does not appear to be a critical factor in snook distribution. However, specific water temperature, water depth, currents and structural habitat components appear to be important in selection of snook nursery habitats.

3.4.5.1 Salinity

Salinity does not appear to be a limiting factor in larval and juvenile snook distribution in the Tampa Bay estuary. Juvenile snook were collected in waters with salinities ranging from 0 to 36 ppt in Tampa Bay (FIMP, unpubl. data). McMichael et al. (1989) reported that juvenile snook (average size = 32 mm SL, range = 10-346 mm), were common in small, quiet marshes, creeks and lagoons, but their presence was not limited to any single salinity range. Larval and juvenile snook are commonly found in both the relatively high salinity waters of lower Tampa Bay (30 ppt) and in the upriver portion of the Little Manatee River (<1 ppt) (Haddad et al. 1992; Peters et al. 1992). Obviously, these fish are tolerant of a wide range of salinities and it appears that absolute salinity has little affect on their distribution in the Tampa Bay estuary. Similar conclusions were drawn for snook inhabiting the Ten Thousand Islands area of Florida. Fore and Schmidt (1973) collected snook (14-196 mm SL) in a wide range of salinities and found no correlation between fish length and salinity.

Perez-Pinzon and Lutz (1991) examined the activity related cost of osmoregulation using juvenile snook (2 months old) which were acclimated to three salinities (freshwater-0 ppt., iso-osmotic/brackish- 12 ppt and seawater- 35 ppt). Snook acclimated to iso-osmotic conditions required less oxygen than fish acclimated to freshwater or saltwater when swimming at the same speed. They suggested that the maintenance of an osmotic gradient in fish of this age demands an appreciable metabolic cost. Juvenile snook had an excellent ability to osmoregulate over a full range of salinities, but when forced to swim at increasing speed, salinity had a marked effect on physiological performance. However, given the large range in salinities in which juvenile snook are found in Tampa Bay, this difference in metabolic cost may not be critical to survival of this species.

Adult snook are euryhaline and generally inhabit brackish water areas where salinities range from 0 to **36** ppt. Adults travel to more saline spawning areas during summer or fall (Thue et al. **1982).**

3.4.5.2 Temperature

Water temperature may influence the temporal periodicity of snook spawning in Tampa Bay. Taylor et al. **(1992)** reported that snook begin to spawn in late spring after water temperatures surpass 27°C.

Juvenile snook in Tampa Bay have been collected in water temperatures ranging from **13.1** to **35.6°C** (McMichael et al. 1989; FIMP, unpubl. data). High temperatures in shallow water during the summer months did not appear to affect habitat utilization by juveniles because large numbers (n= **113)** were collected at **35.6°C** (McMichael et al. 1989).

Low water temperatures, or periodic drops in temperature caused by cold fronts, apparently cause movement out of shallow waters. Juvenile snook collected during cold months were taken from deeper water areas where temperatures appeared to be more stable (McMichael et al. 1989). Many studies have cited cold water temperatures as being limiting factors on snook distribution. In laboratory experiments, mean temperatures at which juvenile snook (125-145 mm TL) stopped feeding was 14.2°C; lost equilibrium was 12.7°C; and died was 12.5°C. Some fish fed at <=2°C above their subsequent lethal temperature. Juvenile mortalities began to occur at and below 14°C (Shafland and Foote 1983). Gilmore et al. (1987) reported adult snook swimming lethargically in water temperature as low as 6.7°C and concluded that they die at temperatures approaching the lower end of the 6-13°C range. Rapid declines in water temperatures can be lethal to snook. Snook have been reported in cold water fish kills in Tampa Bay and other areas of Florida (Storey 1937; Rinckey and Saloman 1964; Taylor, pers. comm. 1992). Water temperatures in Old Tampa Bay and Coffee Pot Bayou, where snook cold mortalities were reported, were 9.6 and 11.8°C, respectively (Rinckey and Saloman 1964). Springer and Woodburn (1960) discuss the incidence of apparent cold mortalities in **Cross** Bayou Canal during 1957, when water temperature was 13°C. Thousands of dead snook were reported throughout Tampa Bay during December, 1989, in areas including Little Manatee River, Manatee River, Bishops Harbor, Lower Tampa Bay, Upper Tampa Bay, and Alafia Rivers (Taylor, pers. comm. 1992). Water temperatures during this period were reported at 9.1 to 10.1°C in Tampa Bay. Anecdotal information suggested that canals and other shallow areas were locations of highest mortality. It appears that although snook can tolerate relatively high seasonal water temperatures, they can be heavily impacted by low water temperatures caused by winter cold fronts.

3.4.5.3 Dissolved Oxygen

Very little information was available on dissolved oxygen requirements of snook in Tampa Bay. Haddad et al. (1992) report that dissolved oxygen values of 4 to 7 ppm were common in areas with snook, although the overall range for DO measurements was from 1.3 to 8.4 ppm. Peterson and Gilmore (1991) reported that low dissolved oxygen conditions could force juvenile snook out of mangrove swamps impounded for mosquito control in Indian River, FL. These authors reported that snook undergo ontogenetic changes in hypoxia tolerance. This pattern was correlated with ontogenic habitat use in east central Florida snook populations (Gilmore et al. 1983). Large snook had more difficulty than small snook in surviving hypoxic conditions in impounded mangroves. Juvenile snook (< 33 mm SL) residing in tidal freshwater or mangrove habitats may be forced to leave these habitats after reaching approximately 100-150 mm as a result of low oxygen conditions (Gilmore et al. 1983). Flooded marsh and mangrove habitats of this type are uncommon in Tampa Bay (McMichael et al. 1989) and similar hypoxic conditions within juvenile snook habitats in Tampa Bay have not been reported. Shafland and Koehl (1979) reported that juvenile snook survived overnight minimum dissolved oxygen concentrations of 0.4 ppm in grow-out ponds, suggesting that they may be able to temporarily withstand low D.O. conditions.

3.4.5.4 Currents

Currents, or near lack of them are thought to be important criteria for larval and juvenile snook nursery areas. Currents measured in nursery habitats ranged from stagnant to about 0.3 m/sec. In general, currents were slow but detectable, and frequently changed their direction of flow either because of their proximity to river and tidal flow or because of wind-driven currents (Haddad et al. 1992; Peters, pers. comm. 1992).

3.4.5.5 Water Depth

Larval and juvenile snook prefer shallow shoreline areas where the depth drops off relatively rapidly to approximately 0.5 m. This type of bottom profile allows cover and structure along the shorelines to remain submerged during low tides, allowing snook to remain within these areas throughout the tidal cycles. In lower Tampa Bay, juvenile snook were found in mangrove fringe areas that remained submerged at a depth of at least 0.3-0.5 m during low tides (Haddad et al. 1992; Peters, pers. comm. 1992). Examples of appropriate and inappropriate snook bottom depth profiles are shown in Figs. 3-10 and 3-11.

3.4.5.6 Structural Habitats

McMichael et al. (1989) stated that it was difficult to label a single habitat in Tampa Bay as primary nursery habitat because relatively large numbers of juvenile snook

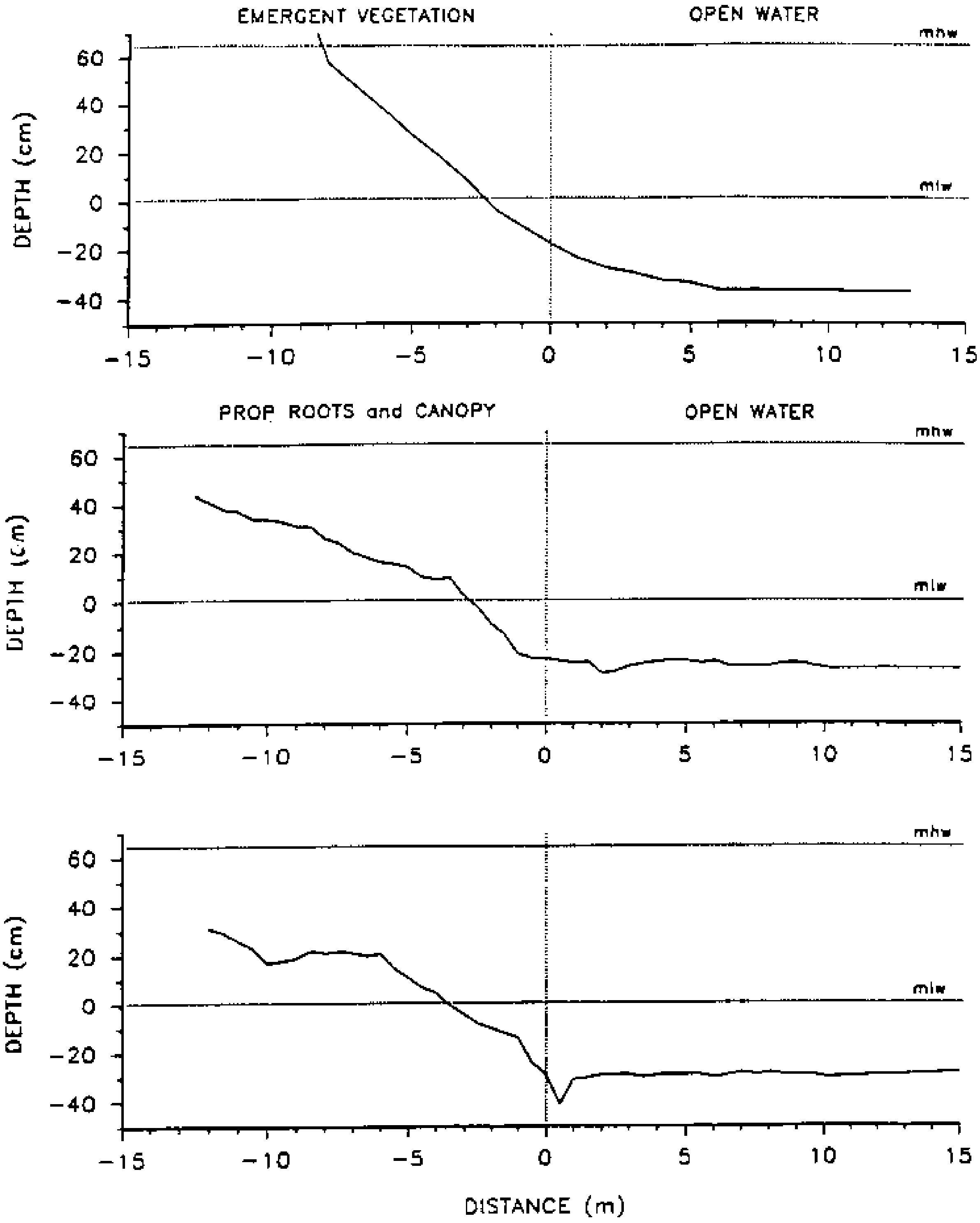

Figure 3-10. Bottom profiles of snook nursery habitat at 11.8 km station (LMR) and two lower Tampa Bay stations. Areas of appropriate snook habitat. Source: Haddad et al. (1992).

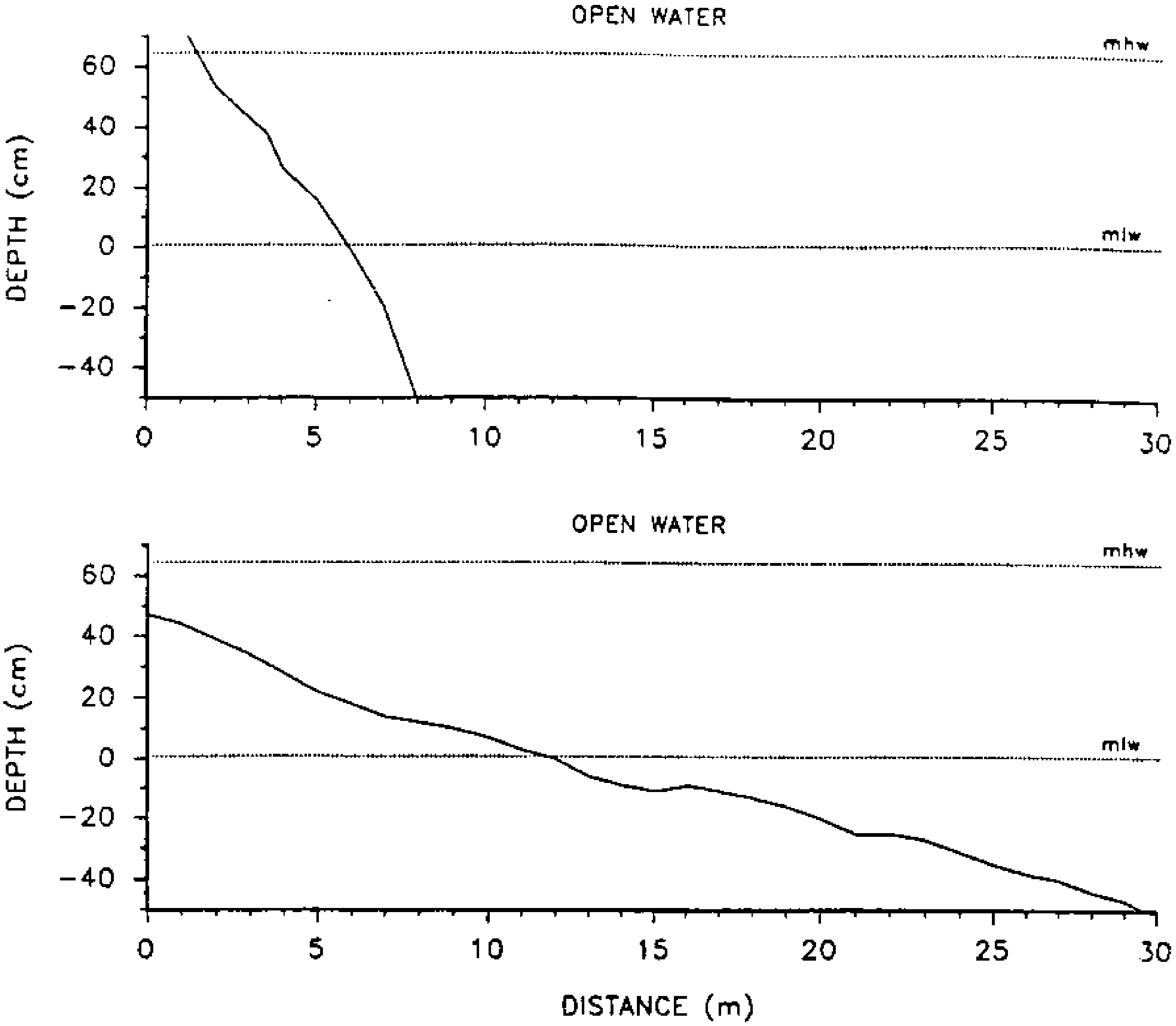

Figure 3-11. Bottom profiles for eroded and depositional bank on Little Manatee River at 5.8 km. Examples of areas inappropriate for snook nursery habitat. Source: Haddad et al. (1992).

were collected in a variety of locations. The majority of juvenile snook collected by these authors (1655 juveniles; 72% 10-70 mm SL) were obtained at stations located in areas characterized as being relatively shallow protected riverine or drainage areas with mud or sand-mud bottom. Primary collection areas included the Alafia River (34%), Cockroach Bay area (23%), Lake Seminole outfall (22%), Little Manatee River (9%) and Bishops Harbor (6%). There were subtle differences in these stations with respect to tidal influence, basin profile, and surface area which affected the amount and rate of salinity and temperature changes in each area. No submerged seagrasses were found at any of these stations, however, various types of other vegetation were present, including floating mats of *Panicum sp., Polygonum sp.* and *Paspalum sp*. Prop roots and rhizomes of red *(Rhizophoramangle)* and black *(Avicenniagerminans)* mangroves were immersed at some stations at high tide, as well as *Juncus sp.* and *Typha sp.* at other stations. Shorelines where most snook were collected had various numbers of mangroves, palms, brazilian pepper and oaks which provided shaded areas for the fish, and often provided physical cover in the form of branches that had fallen into the water or were partially submerged at high tide.

Juvenile snook appear to prefer marginal areas in tidal tributaries rather than the main river channels (Marshall 1958; Edwards 1990; Haddad et al. 1992). These marginal habitats include small tributaries entering the main river, backwater lagoons, tidal creeks, and man-made canals. Juvenile snook entered these low salinity backwater areas in August and September at sizes below 15 mm SL (FIMP 1989). They remained in these areas until at least 130 mm SL.

Larval and juvenile snook also utilize habitat in the higher salinity regions of the Tampa Bay estuary. Peters et al. (1992) reported that **one** of the most productive snook nursery areas were the mangrove swamps of lower Tampa Bay. Dozens of larvae and hundreds of early juveniles were observed among the red mangrove prop roots. Visual censusing indicated that recruitment of larvae into mangrove nursery habitat occurred throughout the summer and fall, at 9-18 day intervals. New recruits arrived in the mangrove fringe at about 7 mm SL and remained in pelagic schools for 2-4 weeks. These authors reported that snook used both high salinity mangrove areas and low salinity marshes as nursery habitats but were more specific in their requirements of other factors such as presence of cover, slow currents and certain water depths.

Haddad et al. (1992) summarized common characteristics of larval and juvenile snook habitats within Tampa Bay. Snook are commonly found in areas of low current and wave motion (resulting in muddy bottoms), shorelines with a relatively steep slope, some type of structure (ranging from mangrove prop roots to floating vegetative mats to branches hanging in the water), and usually shade (e.g. mangrove canopy). Snook are commonly collected in both red mangrove prop roots and *Juncus*/freshwater marsh which ,indicates that the specific type of vegetation or structure also does not limit their distribution. Larval and juvenile snook would not be expected to occur along a shore exposed to the bay or main river flow until they had reached a somewhat larger size (about 100 mm SL). They also would not be expected where the bottom slopes gradually and would cause them to move away from shoreline cover at low tide, nor where the bottom slope drops sharply to a depth of more than 1 m. Also, they would not likely be found in areas completely lacking vegetation or other appropriate structure. These authors reported known and probable snook nursery habitat in the Little Manatee River and Cockroach Bay (Fig. 3-12).

Adult snook are found in rivers, estuaries, and on outer shores of barrier islands, and on nearshore coral reefs (Marshall 1958; Volpe 1959; Fore and Schmidt 1973; Thue et al. 1982; Gilmore et al. 1983). Detailed descriptions of adult snook structural habitat requirements were not available for the Tampa Bay estuary, although Taylor (pers. comm. 1992) reported that principal Tampa Bay snook habitat typically includes mangrove shorelines, freshwater tributaries and nearby access to deep water channels and seagrass beds.

Environmental requirements of snook are summarized in Table 3-6.

3.5 SPOTTED SEATROUT *(Cynoscion nebulosus)*

3.5.1 INTRODUCTION

Spotted seatrout is one of 33 members of the Family Sciaenidae found along the Atlantic, Gulf, and Pacific coasts of the United States (Robins et al. 1980). Spotted seatrout occurs along the Atlantic and Gulf coasts of the U.S. from Cape Cod, Massachusetts to the lower Gulf of Campeche, Mexico, although it is rare in and north of the Delaware Bay (Welsh and Breder 1923; Mather 1952; Tabb 1966; Yanez-Arancibia et al. 1980). It is common throughout the Gulf of Mexico (Pearson 1929; Futch 1970).

Spotted seatrout is an economically and ecologically important species in the Tampa Bay area and throughout the Gulf of Mexico. Spotted seatrout supports valuable commercial and recreational fisheries in the Gulf of Mexico. Economic value of spotted seatrout commercial fisheries displayed marked increases from the early 1950s to early 1980s (Fig. 3-13). Similar economic data are not available for recreational fisheries; however, spotted seatrout is one of the most sought after and most often caught species of sportfish in its range (Tabb 1960). Ecologically, spotted seatrout has been identified as a top carnivore within its ecosystem, and it probably plays a significant role in the structure of estuarine communities (Lassuy 1983).

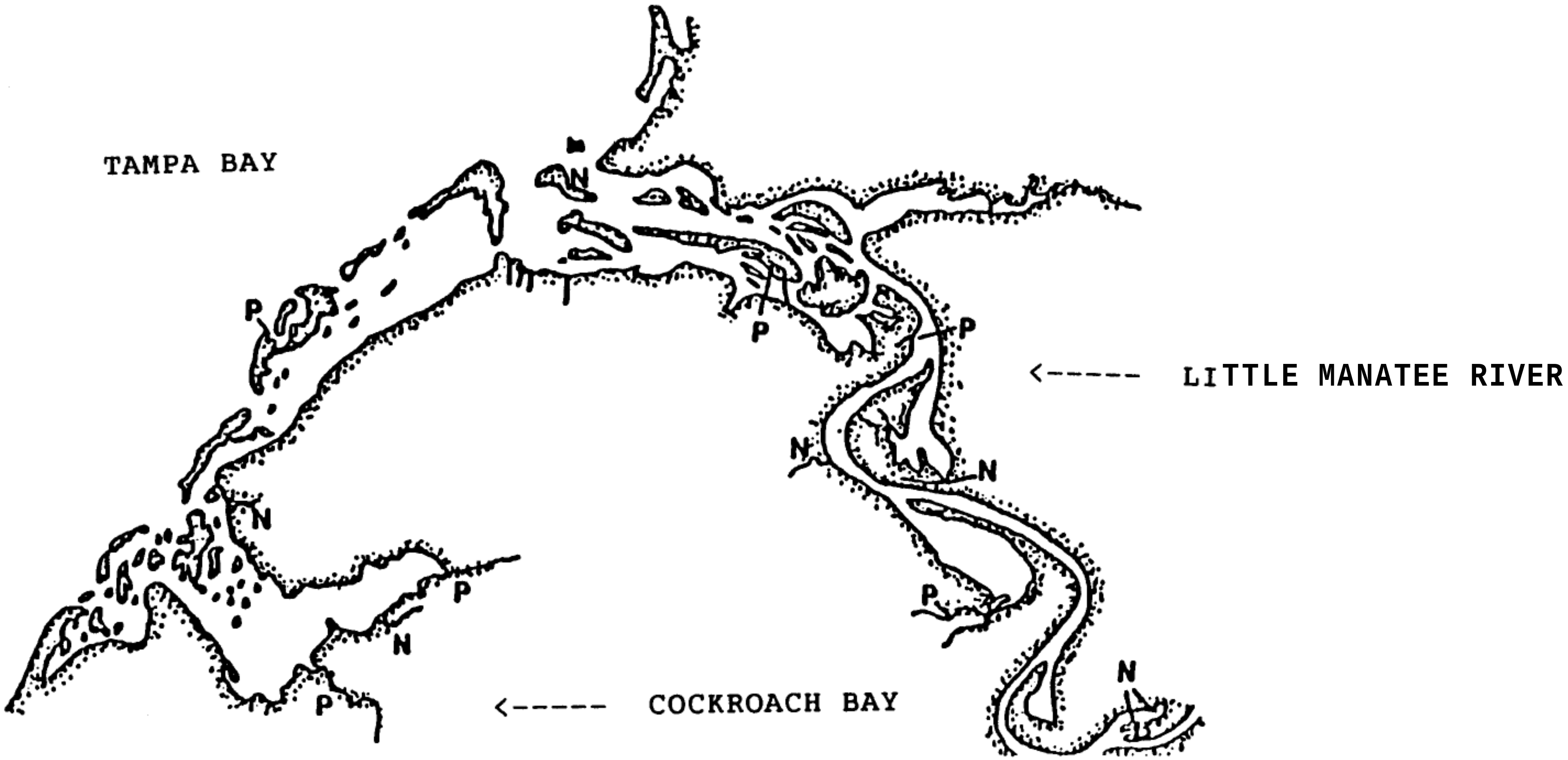

Figure 3-12. Locations of known of snook nursery habitat (N) and probably nursery habitat (P) in the Little Manatee River and Cockroach Bay. Source: Haddad et al. (1992).

Table 3-6. General and preferred ranges and upper and lower tolerance limits for environmental requirements of snook. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Tomperature (°C)	27 (S)				Taylor et al. 1992
				13.1-35.6 (J)	McMichael et al. 1989, FIMP unpubl. data
		9.6. 11.8 (A)			Rinckey and Soloman 1964
		9.1-10.1 (A)			Tavlor pers. comm. 1992
Salinity (ppt)				0-32 (JI	McMichael et al. 1989
				0-36 (J)	FIMP unpubl. data
				0-36 (A)	Thue et al. 1982
Dissolved Oxygen (mg/l)	4-7 (J)			1.3-8.4 (J)	Haddad et al. 1992
		0.4 (J)			Shafland and Koehl 1979
Depth (m)	Shallow shoreline where depth drops rapidly to 0.5 m (L,J)				Haddad et al. 1992
Substrate	mud, sand-mud (J)				McMichael et al. 1989
	Floating vegetation mangrove and structured shoreline (J)				McMichael et al. 1989
	red mangrove prop roots and muddy bottoms (L,J)				McMichael et al. 1989 Peters et al. 1992

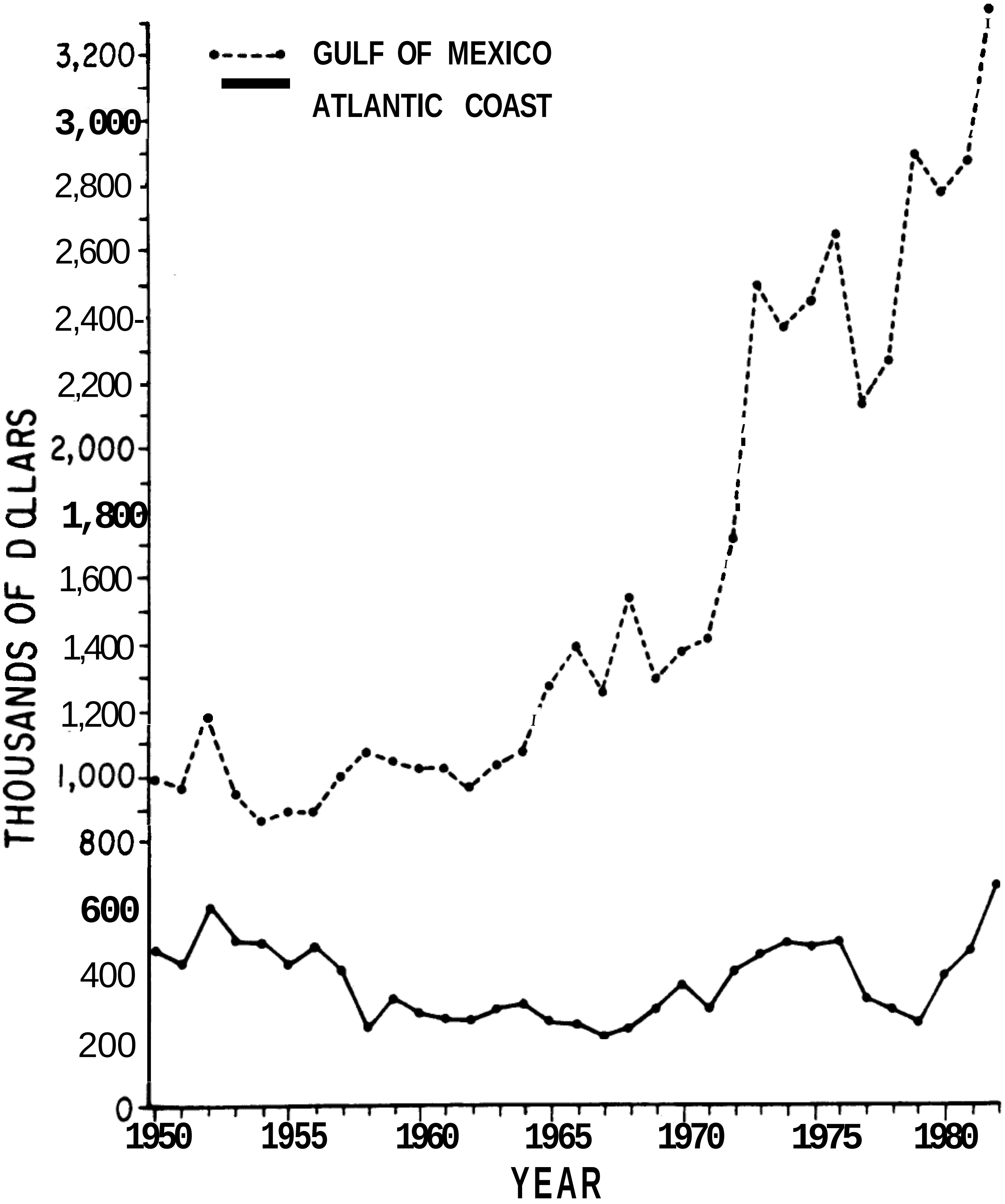

Figure 3-13. Dockside value of U.S. commercial landings for spotted seatrout, 1950-1982 (from Mercer 1984)

3.5.2 LIFE HISTORY

Numerous studies have suggested that spotted seatrout is estuary specific, with very little inter-estuary movement (Moffett 1961; Iverson and Tabb 1962; Iverson and Moffett 1962; Topp 1963; Beaumariage 1964; Beaumariage and Wittich 1966; Weinstein and Yerger 1976). Iverson and Tabb (1962) suggested the existence of subpopulations of spotted seatrout in Florida estuaries, especially those separated by long stretches of coastline.

Spotted seatrout has a protracted spawning season throughout the Gulf of Mexico. Spawning generally occurs from April through October (Pearson 1929; Miles 1950; Moffett 1961; Peebles and Tolley 1988; McMichael and Peters 1989), however it may spawn throughout the year in tropical south Florida (Jannke 1971, Rutherford et al. 1989) and east central Florida (Murphy pers. comm. 1992).

In general, spotted seatrout spawning in Tampa Bay has been well documented. McMichael and Peters (1989) reported that spawning occurs in early April through October with two seasonal peaks during spring and summer. These large peaks were comprised of many smaller peaks, apparently timed with moon phases. Other researchers also documented bi-modal spawning peaks on seatrout in Florida Bay (Jannke 1971, Rutherford et al. 1982), Louisiana (Hein and Shepard 1979), Chesapeake Bay (Brown 1981), and Georgia (Music and Pafford 1984).

The presence of small larvae in mid and lower Tampa Bay suggested that spawning probably occurred in the middle bay to nearshore Gulf waters (McMichael and Peters 1989; Mitchell 1989; Peebles et al. 1992). This agrees with other studies suggesting that seatrout spawn within estuaries (Pearson 1929; Tabb 1966), near passes (Tabb and Manning 1961; Etzoid and Christmas 1979), or outside estuaries (Jannke 1971; King 1971). Spawning location appears to be determined by the characteristics of the local horizontal salinity gradient (Peebles and Tolley 1988). Spotted seatrout larvae were generally more abundant in Tampa Bay than in the more oligohaline Little Manatee River tributary, suggesting that although spawning occurred in Tampa Bay, it did not occur in the riverine portion of the estuary (Peebles et al. 1992). Perret et al. (1980) suggested that the site of spawning may be more related to salinity and temperature than to other water quality parameters. Larvae are typically collected in deep waters in the more central regions of Tampa Bay (McMichael and Peters 1989). Juveniles are found in shallow vegetated seagrass areas and in non-vegetated backwater and oligohaline habitats. Seagrass beds have been documented to be critically important to both juvenile and adult spotted seatrout (Haddad 1989; McMichael and Peters 1989). A generalized spotted seatrout life cycle is presented in Fig. 3-14.

McMichael and Peters (1989) documented age and growth of larval and juvenile spotted seatrout in Tampa Bay. Size at age equations predicted that juveniles will average 15, 35, and 57 mm SL after 1, 2, and 3 months, respectively and will be an average of 84, 114 and 140 mm SL after 4, 5, and 6 months. Age and growth studies on adult spotted seatrout have not been described for Tampa Bay, although they have been

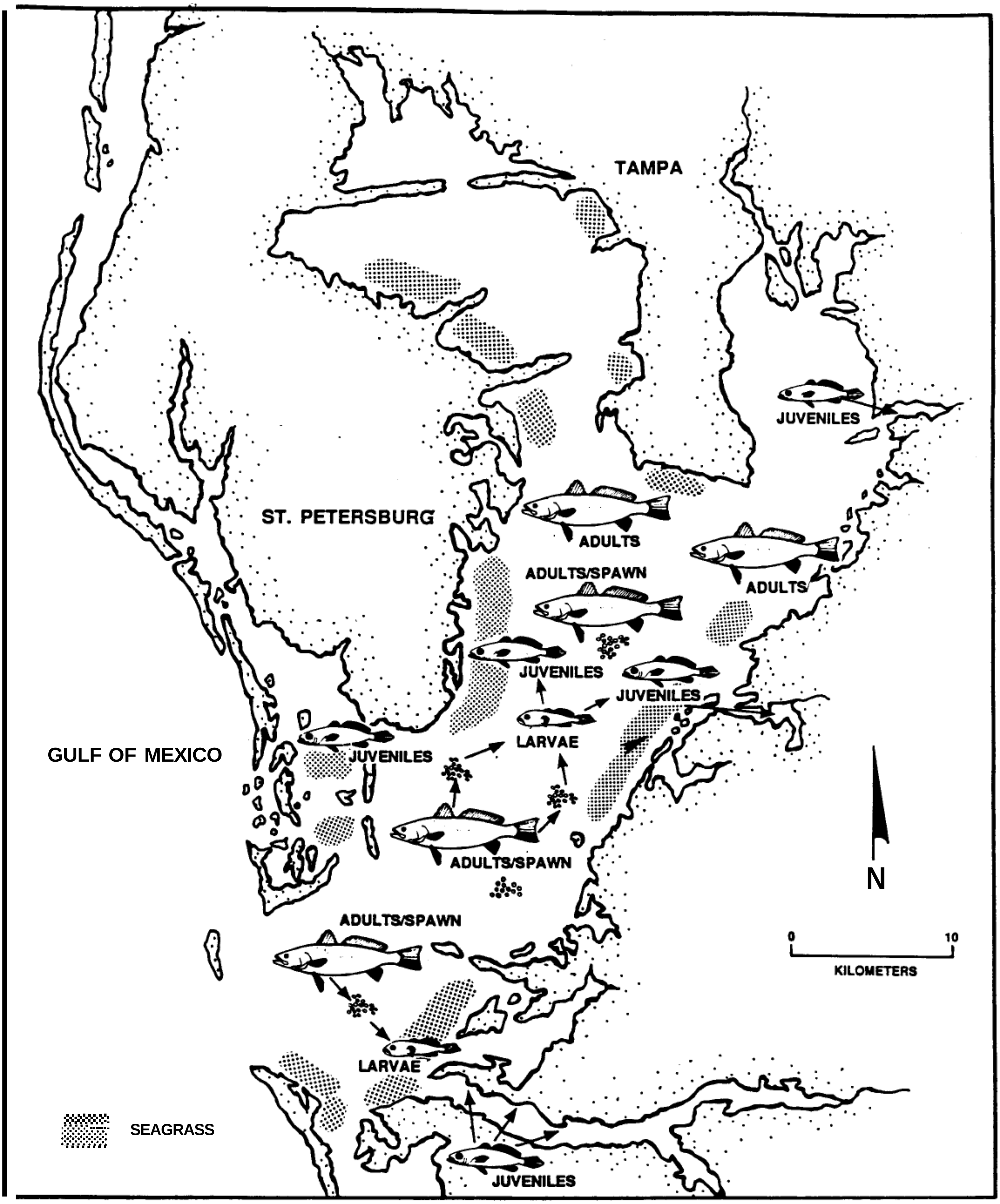

Figure 3-14. Life cycle of spotted seatrout in Tampa Bay

described for many other Florida estuaries such as Charlotte Harbor, Indian River, Apalachicola Bay, and Florida Bay (Murphy, pers. comm. 1992). In general, most spotted seatrout mature between one and three years (Lorio and Perret 1980). Males mature at a smaller size and earlier age than females (Moody 1950; Tabb 1961, Perret et al. 1980). Murphy reported that in Charlotte Harbor, Indian River, and Apalachicola Bay, Florida most males were mature at 305 mm TL. Some fish as small as 229 mm were observed in spawning condition. In all areas, over 95% of observed males were mature by age 1. Fifty percent of females reached maturity by 305 mm, and 90% by 406 mm TL. These sizes corresponded to age 1 and 2.

3.5.3 ECOLOGICAL ROLE

3.5.3.1 Diet

Food habits of spotted seatrout have been fairly well documented throughout its range (Mercer 1984). More than 40 types of prey were identified from stomachs of larval and juvenile spotted seatrout collected in Tampa Bay (McMichael and Peters 1989). These authors found that larval spotted seatrout fed mainly on copepods. These organisms occurred in 83% of the larvae and made up 88% of the total volume. Calanoids were the most frequently found copepod, but harpacticoids and cyclopoids also were present. After the fish reached approximately 45 mm, copepods were no longer eaten. Amphipods and mysids also were eaten by larval and juvenile spotted seatrout, but they made up small percentages in number and volume. Fish and shrimp were the two most important food groups for fish greater than 15 mm (Fig. 3-15). Fish species eaten in Tampa Bay included: bay anchovy, silversides, code goby, clown goby, mojarras, and silver perch. Recent research examining the feeding habits of juvenile (20-90 mm SL) spotted seatrout in Tampa Bay showed similar results (Peebles 1992). The most commonly eaten shrimp was *Hippolyte zostericola;* other species included *Palaemonetes pugio* and *Tozeuma carolinense .* Similar feeding preferences were identified for large juvenile and adult spotted seatrout in other areas of Florida (Moody 1950; Odum 1971; Stewart 1961; Carr and Adams 1973; Tabb 1966; Rutherford et al. 1986; Hettler 1989).

Changes in food habits as a function of growth agree with other studies on spotted seatrout feeding habits (Moody 1950, Tabb 1961, Carr and Adams 1973, Perret et al. 1980, Hettler 1989). Moody (1950) reported that the principal factor affecting the selection of food during growth of seatrout appears to **be** the relative size of the food organism. Tabb (1966) reported that food preferences are probably also influenced by the seasonal availability of prey.

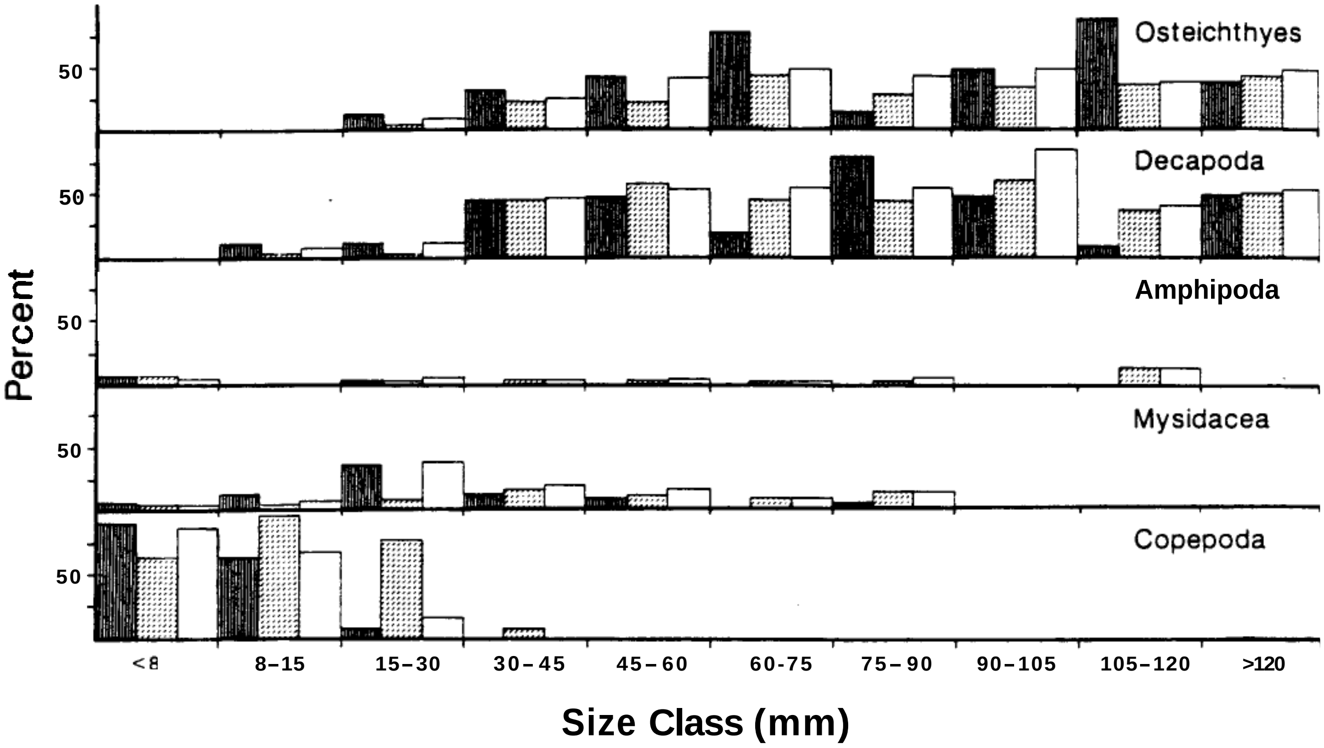

Figure 3-15. Percent volume (shaded), number (stippled), and occurrence (open) of major prey items from 15-mm size classes of *Cynoscion nebulosus* collected in Tampa Bay (from McMichael and Peters 1989)

3.5.3.2 Predators

Nine predators and six competitors of spotted seatrout were listed for Apalachee Bay in northwest Florida (Table 3-71, whereas only five predators were listed in east-central Florida (Klima and Tabb 1959; cited from Mercer 1984). Predators and competitors of spotted seatrout are expected to be similar between Tampa Bay and Apalachee Bay, with the exception of striped bass; and southern rock bass, which are rare in Tampa Bay.

3.5.4 CONTAMINANTS

A few studies have examined contaminants in spotted seatrout. DDT residues were measured in six generations of spotted seatrout from the Laguna Madre, Texas (Butler 1969; Butler et al. 1970; cited from Mercer 1984). Residues reached as high as 8 ppm in the gonads. Gadbois and Maney (1983) determined that the mean level of PCBs in 17 spotted seatrout from the Gulf of Mexico was 0.16 ppm. This level was below existing (5 ppm) or proposed (2 ppm) maximum permissible in food fish.

3.5.5 ENVIRONMENTAL REQUIREMENTS

Tabb (1958) listed environmental requirements of spotted seatrout that he felt were most important in determining the abundance and success of seatrout in Florida. These included: 1) large areas of shallow, quiet, brackish waters; 2) extensive grassy areas dominated by turtle and shoal grass; 3) areas of 3 to 6 m depth adjacent to grass flats to be used for refuge from winter cold; 4) an abundant food supply; 5) absence of predators; 6) absence of competitors; and 7) temperature ranging between 15° and 27°C.

The Tampa Bay estuary provides many of the water quality and structure conditions required by spotted seatrout throughout its life history. Water quality parameters include salinity, temperature, dissolved oxygen and turbidity. Structural habitat includes water depth, vegetation, substrate and morphology. Environmental requirements of spotted seatrout will be discussed for each of these topics.

3.5.5.1 Salinity

Tolerance to a broad range of salinities is important for estuarine species such as spotted seatrout, which are likely to experience large salinity fluctuations. Much of the data available for salinity preferences of spotted seatrout relate to spawning, larval, and early juvenile life stages. Spotted seatrout spawn in a wide range of salinities, although optimum salinities determined from lab experiments have been reported to be 20 to 35 ppt (Arnold et al. 1976). Taniguchi (1980) reported that optimum salinity for eggs and larvae is 28.1 ppt; however, 100% survival is predicted between 18.6 and 37.5 ppt. Banks et al. (1991) reported that spotted seatrout larvae showed a greater ability to adapt to

Table 3-7. List of predators and competitors of spotted seatrout in Apalachee Bay in northwest Florida (from Klima and Tabb 1959)

CommonName	Scientific Name	Relation	Occurrence
Striped bass	***Morone saxatilis*** (Walbaum)	Predator (?)	Resident
Snook	***Centropomus undecimalis*** (Bloch)	Predator	Seasonal
Tarpon	***Megalops atlantica*** Valenciennes	Predator	Seasonal
Alligator gar	***Lepisosteus spatula*** Lacepede	Predator(?)	Seasonal
Sea catfish	***Galeichthys felis*** (Linnaeus)	Competitor	Resident
Barracuda	***Sphyraena barracuda*** (Walbaum)	Predator	Occasional
Spanish mackerel	***Scomberomorus maculatus*** (Mitchill)	Predator	Seasonal
King mackerel	***Scomberomorus cavalla*** (Cuvier)	Predator	Seasonal
Bluefish	***Pomotomus saltatrix*** (Linnaeus)	Predator	Resident
Grouper	***Mycteroperca sp.***	Competitor	Resident
Silver perch	***Bairdiella chrysoura*** (Lacepede)	Predator and competitor	Resident
Red drum	***Sciaenops ocellatus*** (Linnaeus)	Competitor	Resident
spot	***Leiostomus xanthurus*** (Lacepede)	Competitor	Resident
Croaker	***Micropogon undulatus*** (Linnaeus)	Competitor	Resident
Southern rock bass	***Ambloplites rupestris ariomus*** Viosca	Competitor	Resident

brackish or lower than normal salinities than to hypersaline conditions. This would be an obvious benefit in the estuarine environment where salinities are more likely to fluctuate rapidly towards brackish conditions; e.g., large storms can have a more immediate effect than gradual hypersalinity caused by drought and evaporation. In Florida Bay, seatrout post larvae were collected between 8 and 40 ppt (mean=33.2), and these values placed spotted seatrout among the most euryhaline of the sciaenid larvae (Rutherford et el. 1986). Salinities observed at stations in Tampa Bay where larval spotted seatrout were collected during spawning season are consistent with optimal salinities reported from lab experiments; salinities recorded in the mid and lower bay were 18.5 to 30.5 ppt and 23 to 36.0 ppt, respectively (McMichael and Peters 1989).

Research conducted in Tampa Bay suggests that juvenile spotted seatrout are extremely euryhaline. McMichael and Peters (1989) collected juvenile spotted seatrout in seagrass beds *(Thalassia, Halodule* and *Syringodium)* where salinities ranged from 16 to 35 ppt (mean=26 ppt.) and in backwater areas where salinities ranged from 0 to 27 ppt (mean=7 ppt).

Recent research on the life history of juvenile spotted seatrout suggests that tidal tributaries may be important nursery areas (McMichael and Peters 1989; Edwards 1990; Haddad et al. 1992; Peebles et al. 1992). Juvenile seatrout in the Manatee River were collected at stations with salinities ranging from 0 to 22 ppt (Edwards 1990). Over 90% of the 221 spotted seatrout collected in that study were collected at salinities less than 12 ppt, and over 62% were collected where salinities were less than 7 ppt; however, the salinities in the river during this study were very low, and the author felt it was not appropriate to interpret these salinities as preferred or optimal for seatrout. His general conclusion was that spotted seatrout juveniles were capable of using a broad range of salinities within the Manatee River estuarine system; however, they preferred intermediate salinities to extremely low salinities. A similar conclusion was proposed for spotted seatrout in the Little Manatee River. Spotted seatrout were abundant as larvae and juveniles in the Little Manatee River area, but peak abundances occurred in moderate salinities nearer the mouth of the river, generally between kilometers 0 and 4 (Peebles et al. 1992; Haddad et al. 1992, Fig. 3-16). They were most abundant in the polyhaline (> 18 ppt) and mesohaline (5-18 ppt) reaches of the river, but moderate catches were reported in the oligohaline (0.5-5 ppt) and freshwater reaches of the tributary (Fig. 3-17). Catch rates in a hot/high salinity year (1990) were lower than in other years, which was opposite of what might be expected if salinity alone controlled the distribution and abundance of this species in the Little Manatee River (Haddad et al. 1992). Although it appears that tidal tributaries provide important nursery areas for spotted seatrout, they prefer areas with more intermediate salinities to those with extremely low salinities.

Adult spotted seatrout are euryhaline and have been collected in waters where salinities ranged from 0.2 to 70 ppt (Simmons 1957). Abrupt declines in salinity (changes of 10-15 ppt), such as those caused by large storms or hurricanes, may result in mass migrations of seatrout out of the estuary (Tabb 1966). It has been suggested that mortality may occur at salinities less than 5 ppt (Kostecki 1984). Wohlschlag and Wakeman (1978) found optimal salinity levels for spotted seatrout were on the order of

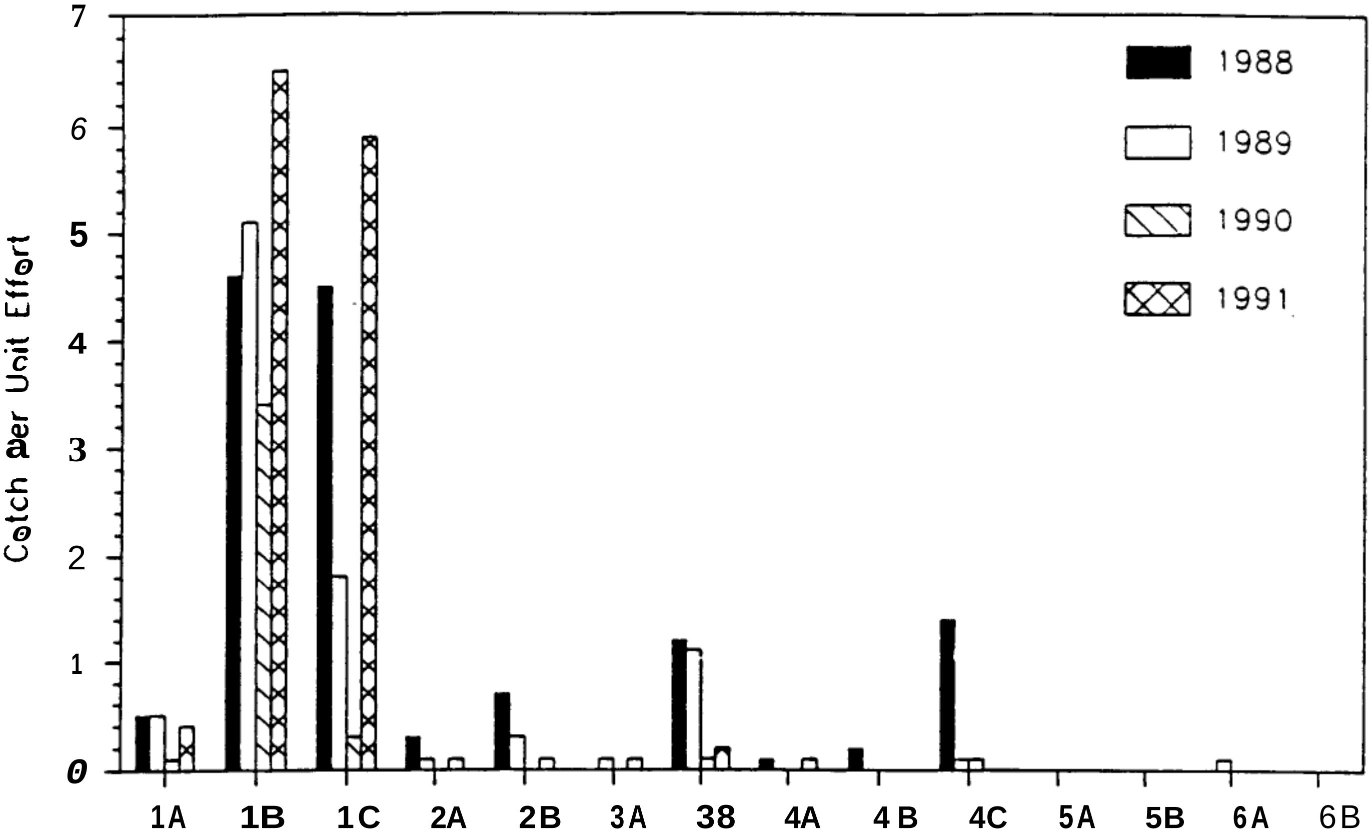

Figure 3-16. CPUE (# fish/100 m²) for juvenile spotted seatrout by location. Station 1 = rkm 0, station 2 = rkm 2.5, station 3 = rkm 5.8, station 4 = rkm 11.8, station 5 = rkm 15.5, station 6 = rkm 16.4. Source: Haddad et al. 1992.

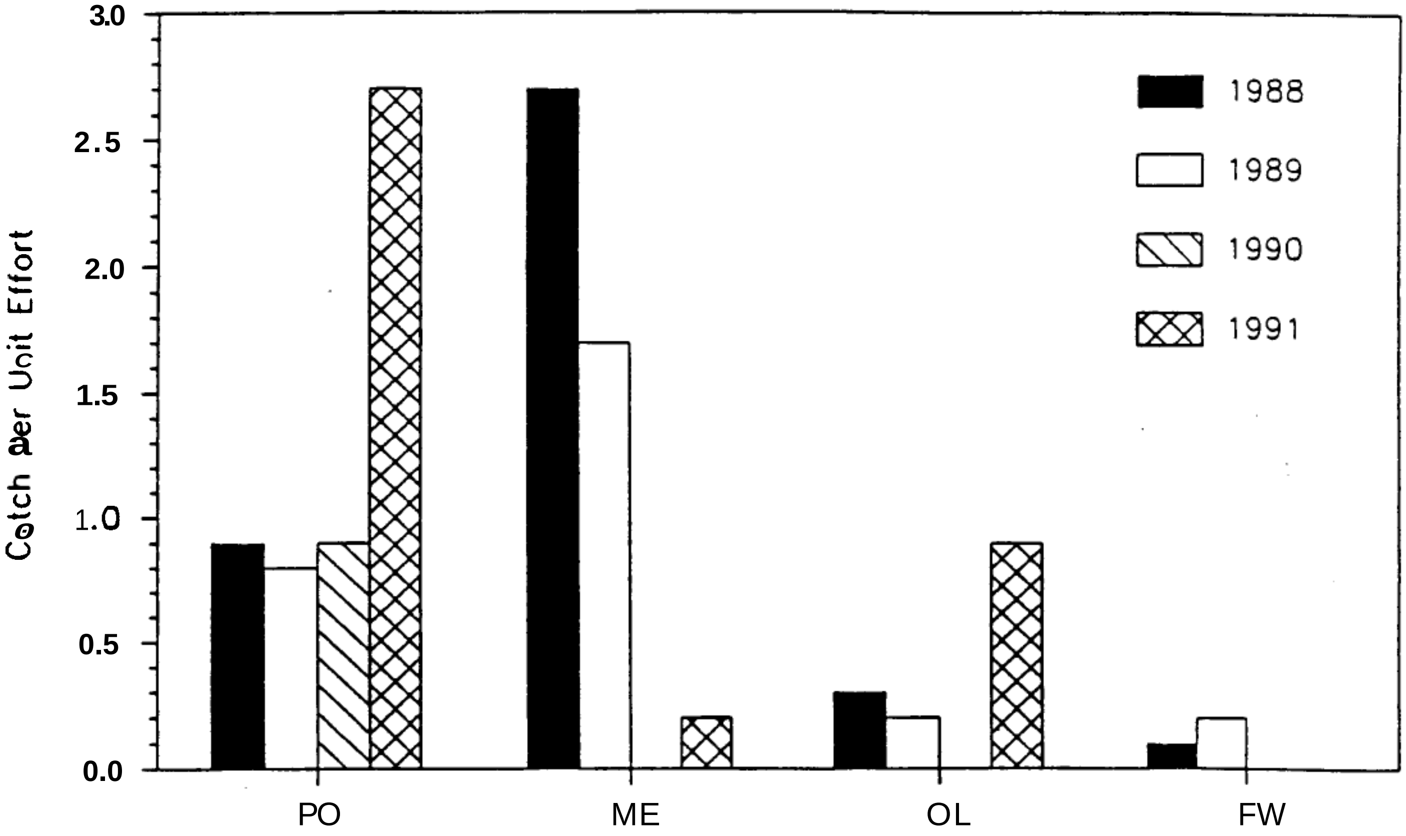

Figure 3-17. CPUE (# fish/100 m²) for juvenile spotted seatrout by salinity region. PO = polyhaline (> 18 ppt), ME = mesohaline (5-18 ppt), OL = oligohaline (0.5-5 ppt), and FW = freshwater (0-0.5 ppt). Source: Haddad et al. 1992.

20 ppt at 28°C. They reported rarely collecting seatrout below 10 ppt or above 45 ppt in south Texas waters.

3.5.5.2 Temperature

Seatrout spawning reportedly occurs in many estuaries in temperatures of 21.0° to 28.3°C (Kostecke 1984). Taniguchi (1980) reported an optimal temperature of 28°C for eggs and larvae but predicted 100% survival between 23.1° and 32.7°C. Surface water temperatures in mid and lower Tampa Bay during seatrout spawning months (April through October) were 17.5° to 31.0°C and 15.5° and 30.5°C, respectively (McMichael and Peters 1989). Juvenile spotted seatrout were collected in Tampa Bay in water temperatures ranging from 14.1 to 29.7°C (FIMP, unpubl. data).

Spotted seatrout adapt to natural temperature fluctuations by moving from shallow areas to deeper channels within the estuary or offshore (Tabb 1966). Water temperatures of 16° to 27°C are suitable for adult spotted seatrout, whereas temperatures as low as 7° to 10°C have been identified as detrimental (Tabb 1966). Simmons (1957) reported that in Laguna Madre, Texas seatrout actively feed at temperatures between 4o and 33°C; however, other studies suggest that extremely cold water temperatures can be fatal to spotted seatrout. Mass mortalities may occur during cold periods when water temperatures drop to 4o to 7°C (Gunter 1941; Gunter and Hildebrand 1951; Tabb 1958; Moore 1976). Seasonal cold fronts can cause rapid temperature declines in shallow estuarine waters. Buckley (1984) suggested that the rate of temperature decline is more important than absolute temperature in influencing cold water survival of red drum. A similar situation may occur for spotted seatrout which move to deeper areas of the bay during winter months (Pearson 1929; Tabb 1966). These areas may act as a refuge from rapid temperature declines. In Georgia estuaries, spotted seatrout were observed to move into deeper waters at temperatures greater than 25°C or less than 16°C (Mahood 1975). No juveniles were collected during the winter months in the Little Manatee River area (Haddad et al. 1992). Water temperature may also be an important factor limiting growth and production of spotted seatrout (Tabb 1966).

3.5.5.3 Dissolved Oxygen

There are no data relating the distribution of spotted seatrout to DO concentrations (Mercer 1984). Seatrout were reported in fish kills caused by oxygen depleted waters have been reported in Mississippi (Etzoid and Christmas 1979). Vetter (1982) examined the metabolic rate of spotted seatrout in relation to temperature changes in Redfish Bay, TX. Seasonal variation in metabolic rate ranged from 123 mg O_2/kg/hr at 3O°C to 49 mg O_2/kg/hr at 15°C. These rates were significantly lower at 3O°C and significantly greater at 15°C and 2O°C than for the sympatric sand seatrout *(C. arenarius)*. The greater metabolic compensation of spotted seatrout may be an adaptation to year round exploitation of the estuarine habitat, which experiences more extreme temperatures than offshore waters. Sand seatrout rely more heavily on migration into and out of the estuary to avoid temperature extremes (Vetter 1982).

3.5.5.4 Turbidity

Very little information is available relating to turbidity tolerance of spotted seatrout. Pearson (1929) suggested that spotted seatrout preferred areas of low turbidity. Also, some mortalities in estuaries have been attributed to high turbidity resulting from the influence of a hurricane (Tabb and Manning 1961; Perret et al. 1980).

3.5.5.5 Water Depth

Pearson (1928) reported that spotted seatrout spawned in bays and lagoons at depths of 3 to 4.5 m, whereas Tabb (1966) observed spawning in deep holes and channels of estuaries. McMichael and Peters (1989) found that the vertical and areal distribution of larval seatrout in mid and lower Tampa Bay showed no consistent pattern. Many juvenile spotted seatrout were collected in shallow (< 2 m) seagrass and backwater areas in Tampa Bay (McMichael and Peters 1989). as well as in shallow oligohaline habitats (Edwards 1990; Peebles et al. 1992). Tabb (1966) reported numerous early juvenile seatrout in deep channels; however, in Tampa Bay, no early juveniles were collected in the bottom of the deep (12 m) ship channel, although two other sciaenid species *(Cynoscion arenarius* and *Menticirrhus americanus)* of this size were found (McMichael and Peters 1989). Much of the commercial and recreational catch of adult spotted seatrout comes from shallow seagrass areas (Haddad 1989).

3.5.5.6 Structural Habitat

In general, seagrass beds appear to be the preferred habitat for postlarval, juvenile and adult spotted seatrout (Pearson 1929; Moody 1950; Springer and Woodburn 1960, Tabb 1966; McMichael and Peters 1989; Rutherford et al. 1989). However, they may also occur abundantly near areas without extensive seagrass beds (Lorio and Perret 1980; McMichael and Peters 1989; Edwards 1990). Chester and Thayer (1990) believe that seagrass meadows with mixtures of *Thalassia testudinum* and either *Halodule wrightii* or *Syringodium filiforme* are critical habitats for spotted seatrout in Florida Bay. They suggested that the abundance and distribution of juvenile spotted seatrout appeared to be influenced by biomass, shoot density, and species composition of the seagrass community. Juvenile seatrout remain in the submerged vegetation during summer but may move to deeper water as the water cools in the winter (Pearson 1929, Miles 1950, Moody 1950).

McMichael and Peters (1989) described the early life history of spotted seatrout in Tampa Bay. Juvenile spotted seatrout use seagrass beds (*Thalassia, Syringodium, Halodule)* as their primary habitat, but they were also found in unvegetated backwaters and oligohaline habitats. The seagrass beds where seatrout were collected occurred throughout the middle and lower bay along both exposed and semi-protected shorelines with sand or sand-mud bottoms. Backwater areas containing seatrout were usually in quiet or slow-moving, fresh-water influenced rivers, bayous, and tidal creeks that had mud or sand-mud bottoms. Seventy-eight percent of all juveniles were collected over seagrass, although less than 40% of collections were made at seagrass habitats, underscoring the importance of these habitats to juvenile seatrout. Similar size distributions in both seagrass and backwater habitats suggests recruitment of larvae into both areas and little or no size-related movement between them. Importance of oligohaline habitats is described in the salinity section of this report.

Declines in seagrass habitat have been linked with declining seatrout populations in Tampa Bay. Lewis et al. (1985) estimated an 81% loss of seagrasses in Tampa Bay since the 1800s. Seagrass meadows in the lower-salinity areas of the bay (Hillsborough Bay, upper Old Tampa Bay) have disappeared largely in recent years; thus, the primary nursery area for this species may have been reduced severely. The major decline in the commercial catch of spotted seatrout in the bay (Lombardo and Lewis 1985) may be a result of this loss of nursery habitat. Haddad (1989) emphasizes the importance of seagrass beds to all age classes of spotted seatrout and documents declines in seatrout abundance and commercial harvest that may have resulted from the declining seagrass beds. He reports a 50% decline in Tampa Bay seagrasses between the 1950s and 1982.

Spotted seatrout historically have provided an important recreational and commercial fishery in the Tampa Bay region. Commercial harvest records for Tampa Bay show marked declines between 1950 and the early 1980s (Fig. 3-18). Scientific data documenting the reasons for the decline of this species do not exist. Based on existing knowledge of the use of seagrass beds by juvenile and adult seatrout in the Tampa Bay system, and the fact that seatrout tend to be non-migratory (Moffett 1961; Iverson and Tabb 1962), the estuary can be assumed to produce and support its own population with minimal external influences. Although numerous factors can control spotted seatrout population, a loss of 50% to 80% of the Tampa Bay seagrasses probably would affect landings. It was also assumed that with the loss of seagrasses, the actual production potential (carrying capacity) of this species would be reduced in the bay, and the seatrout population could not recover to historical levels, even if fishing pressures were eliminated (Haddad 1989).

Sykes and Finucane (1965) report that areas such as Hillsborough Bay have been heavily impacted by industrial and domestic wastes. Lewis and Estevez (1988) indicated that seagrass meadows in these regions have largely disappeared in recent years, and that this may be contributing to the loss of these low salinity areas as nursery grounds. Very few spotted seatrout were collected in this region relative to other areas in Old Tampa Bay and in the lower and central portions of Tampa Bay (Sykes and Finucane 1965). In McKay Bay, an embayment of Hillsborough Bay, only one spotted seatrout (of 23,740 fish) was collected between 1977 and 1979 (Price and Schlueter 1985).

Environmental requirements of spotted seatrout are summarized in Table 3-8.

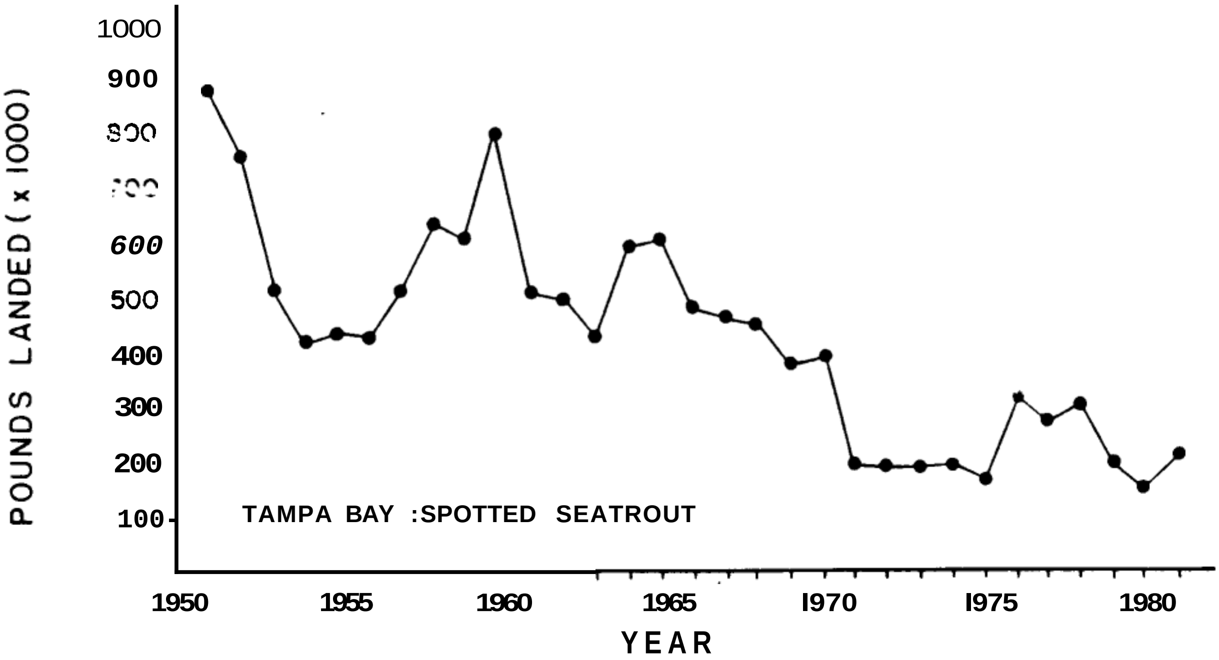

Figure 3-18. Landings of spotted seatrout in Tampa Bay (from Haddad 1989)

Table 3-8. General and preferred ranges and upper and lower tolerance limits for environmental requirements of spotted seatrout. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				21-28 (S)	Kostecke 1984
	28 (E,L)			23-32.7 (E,L)	Taniguchi 1980
				14.1-29.7 (J)	FIMP (unpubl. data)
	16-27 (A)	7-10 (A)			Tabb 1966
		4-7 (A)			Gunter 1941; Gunter and Hildebrand 1951; Tabb 1958; Moore 1976
Salinity (ppt)	20-35 (S)				Arnold et al. 1976
	28.1 (E,L)			18.6-37.5 (E,L)	Taniguchi 1980
				8-40 (L)	Rutherford et al. 1986
				0-35 (J)	McMichael and Peters 1989
	>5. (J)				Peebles et al. 1992, Haddad et al. 1992
				0.2-70 (A)	Simmons 1957
	20 (A)	10 (A)	45 (A)		Wakeman 1978
		<5 (A)			Kostecki 1984
Depth (m)	<2 (J)				McMichael and Peters 1989; Edwards 1990; Peters et al. 1992
Substrate	seagrass (L,J,A)				Springer and Woodburn 1960; McMichael and Peters 1989; Rutherford et al. 1989
				unvegetated - vegetated	Lorio and Perret 1980; McMichael and Peters 1989

3.6 RED DRUM *(Sciaenops ocellatus)*

3.6.1 INTRODUCTION

Red drum is an estuarine-dependent sciaenid found in nearshore waters along the Atlantic coast from the Gulf of Maine to Key West, FL (Yokel 1966; Lux and Mahoney 1969; Ross et al. 1983), although it is only occasionally found north of Chesapeake Bay (Yokel 1980). In the Gulf of Mexico, red drum range from south Florida north and west along the Gulf coast into northern Mexico (Swingle et al. 1984).

In the United States, red drum support important recreational and commercial fisheries in many coastal areas of the south Atlantic and Gulf of Mexico (Mercer 1984; Buckley 1984). During 1980 to 1984, approximately 75% of Florida's inshore commercial red drum harvest came from southwest Florida (Edwards 1990). In recent years, red drum stocks in the Gulf of Mexico declined drastically (Goodyear 1989). In response to these declines, management regulations enacted in Florida during 1987 prohibited the sale of red drum (Taylor, pers. comm. 1992). This measure eliminated the commercial harvest of this species in Florida waters.

3.6.2 LIFE HISTORY

In the Gulf of Mexico, red drum spawn in nearshore waters, in passes and inlets, and inside large estuaries (Pearson 1929; Yokel 1966; King 1971; Jannke 1971; Perret et al. 1980; Swingle et al. 1984; Peters and McMichael 1987; Murphy and Taylor 1990). Most spawning occurs in late summer and fall in the Gulf of Mexico (Pearson 1929; Miles 1951; Springer and Woodburn 1960; Mercer 1984).

Red drum spawning in Tampa Bay occurs from mid-August through mid-November, with peaks in late August through mid-October (Peters and McMichael 1987; Murphy and Taylor 1990). Murphy and Taylor (1990) presented evidence suggesting that red drum spawning near Tampa Bay occurs in the nearshore Gulf of Mexico, in the vicinity of passes and within the Tampa Bay estuary. Red drum collected large distances (42 km) inside Tampa Bay had post ovulatory follicles which were suggestive of recent spawning, potentially inside the estuary. Spawning peaks appeared to correspond to new or full moons, which suggested that tidal influence, rather than actual moon phase may affect spawning time (Peters and McMichael 1987).

Red drum larvae are carried through inlets and passes into estuaries by tidal currents (Pearson 1929; Yokel 1966; Jannke 1971; Holt and Arnold 1982). Yokel (1966) reported that red drum larvae (1.5 to 7.0 mm) are generally found in the open gulf or short distances within the estuary. Ichthyoplankton samples collected at discrete depths off the Sunshine Skyway suggested that red drum larvae preferred bottom to mid-depth waters (Robison 1985). Considering the circulation patterns within Tampa Bay, a larval fish may have a greater probability of achieving landward transport and/or estuarine retention by remaining near bottom waters in all tidal phases (Robison 1985). Thus, the affinity of larval red drum to bottom waters may be correlated with a greater degree of dependency on the estuary nursery grounds of upper Tampa Bay. Peebles (pers. comm. 1992) reports that distribution of larval red drum in the water column may also be influenced by time of day and size of larvae.

Red drum larvae become less abundant, but larger in size proceeding from the mouth of Tampa Bay to the upper bay, suggesting that the majority of spawning takes place in the bay mouth or nearshore Gulf waters (Peters and McMichael 1987). The majority of larvae collected in plankton tows made by Peters and McMichael (1987) were from the lower (Sunshine Skyway) and middle bay areas. Red drum larvae collected with seines were also found primarily at lower (Big Bayou) and mid-bay (northern Boca Ciega Bay) stations. Peters and McMichael (1987) found juveniles in a wide variety of habitats, and they proposed that in large, open bay estuaries such as Tampa Bay, larvae hatched near the bay mouth may enter the estuary and settle out along the various bay shores as small juveniles before concentrating in backwater nursery areas. Juveniles reportedly leave these shallow areas for deeper bays and bayous as their size increases (Peters and McMichael 1987). Very little information has been reported on the distribution of adult red drum in Tampa Bay. A generalized life cycle of red drum in the Tampa Bay estuary is shown in Fig. 3-19.

Juvenile red drum grow rapidly and reach approximately 300 mm SL after one year of growth (Peters and McMichael 1987). Tampa Bay red drum continue to grow rapidly until age four or five, when growth slows markedly (Murphy and Taylor 1990). Average observed sizes of fishes aged 1 to 3 were not significantly different between sexes, although Murphy and Taylor (1990) identified large differences in age of maturity between female and male red drum in Tampa Bay. Males matured at smaller sizes and younger ages than did females. Some males were sexually mature at 400 mm length. Lengths at 50% maturity were 529 mm. Most males mature at age one or two and all were mature by age 3. Some female red drum were mature after 600 mm, with 50% mature by 825 mm. Some females were mature at age three, and all were mature at age six. Maximum age observed for Tampa Bay red drum was 24 years old.

Predicted red drum length at a particular age was greater on the Atlantic coast of Florida (Indian River) than for Tampa Bay. Florida estimates of asymptotic length were generally greater than other reported values from Texas bays and Mississippi Sound, suggesting that red drum grow larger in Florida or that larger fish, which predominantly inhabit continental shelf waters, were not adequately sampled in Texas and Mississippi (Murphy and Taylor 1990).

It appears there is very little interbay movement of sub-adult red drum in Florida. Tagging studies in Florida estuaries revealed that greater than 85% of recaptured red drum moved less than 10 km from their tagging site (Ingle et al. 1962; Beaumariage and Wittich 1966; Beaumariage 1969). Along the Gulf coast, adult red drum move from the estuaries to the Gulf of Mexico at maturity (> 700 mm FL)(Yokel 1966). In the Gulf of Mexico, adult red drum tend to travel in schools close to shore until summer, when some move

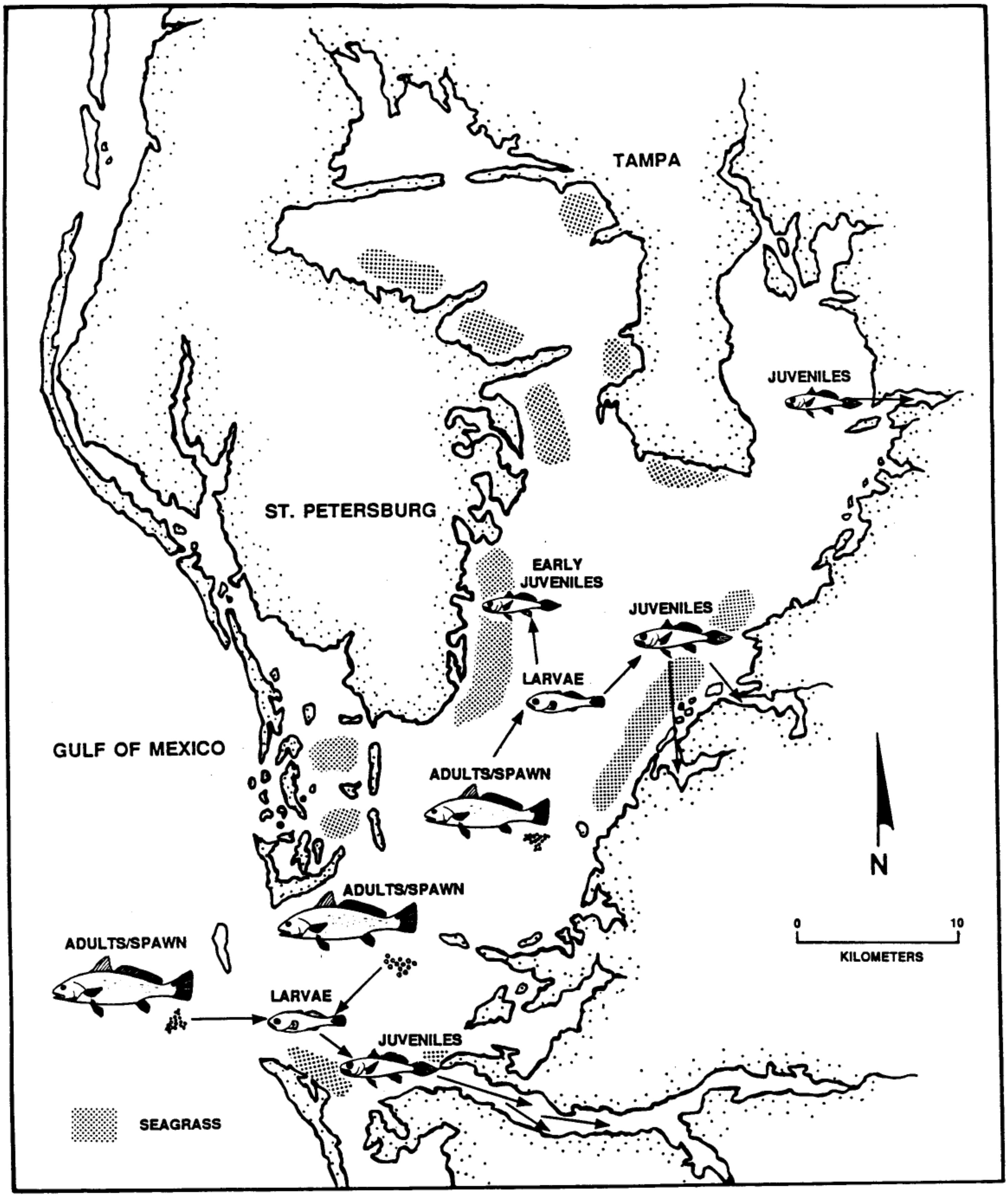

Figure 3-19. Red drum life cycle in the Tampa Bay estuary

into estuaries (Pearson 1929); however, some of the larger fish remain in the Gulf of Mexico year round (Simmons and Breuer 1962; cited from Reagan 1985).

3.6.3 ECOLOGICAL ROLE

Fish, shrimp and crabs appear to constitute primary prey for red drum ranging from 50 to 1000 mm, and the relative abundance of these food items where red drum are present will greatly affect both growth and survival (Swingle et al. 1984). Approximately 50 prey items were identified from stomachs of larval and juvenile red drum in the Tampa Bay estuary (Peters and McMichael 1987). Copepods were the major prey of larvae, with cyclopoids, calanoids, and harpacticoids found in decreasing orders of abundance. Juveniles greater than 45 mm had no copepods in their stomachs, but mysids were abundant food items for all size classes greater than 8 mm. Small juvenile red drum fed predominantly on mysids, amphipods and polychaetes, while larger juveniles fed on fish, crabs and shrimp (Fig. 3-20). Other studies also reported changes in diet with increasing size (Bass and Avault 1975; Yokel 1966). Small juveniles collected over seagrass and non-seagrass areas had similar diets, although minor differences were found in sedentary prey organisms or those with strict habitat requirements, (e.g., polychaetes only in stomachs from unvegetated areas) (Peters and McMichael 1987).

Adult red drum feed primarily on macroinvertebrates and fishes. Boothbay and Avault (1971) provided detailed descriptions of adult red drum feeding habits in Louisiana (Table 3-9). Descriptions of adult red drum feeding habits were not available for the Tampa Bay estuary.

3.6.3.1 Predators

Red drum are major predators in estuaries but their role as prey has not been well documented (Springer and Woodburn 1960). They are potential prey of larger piscivorous fish, as well as to larger red drum.

3.6.3.2 Biological Impacts

Red drum mortalities resulting from red tide outbreaks have been observed. Ptychodiscus brevis, a toxic dinoflagellate caused mortality in hundreds of red drum in Tampa Bay during the summer of 1971 (Steidinger pers. comm.; cited from Swingle et al. 1984).

3.6.4 CONTAMINANTS

No data was available to document contaminant concentrations in red drum from the Tampa Bay estuary. Some data on trace metals and PCB's exists for other estuarine

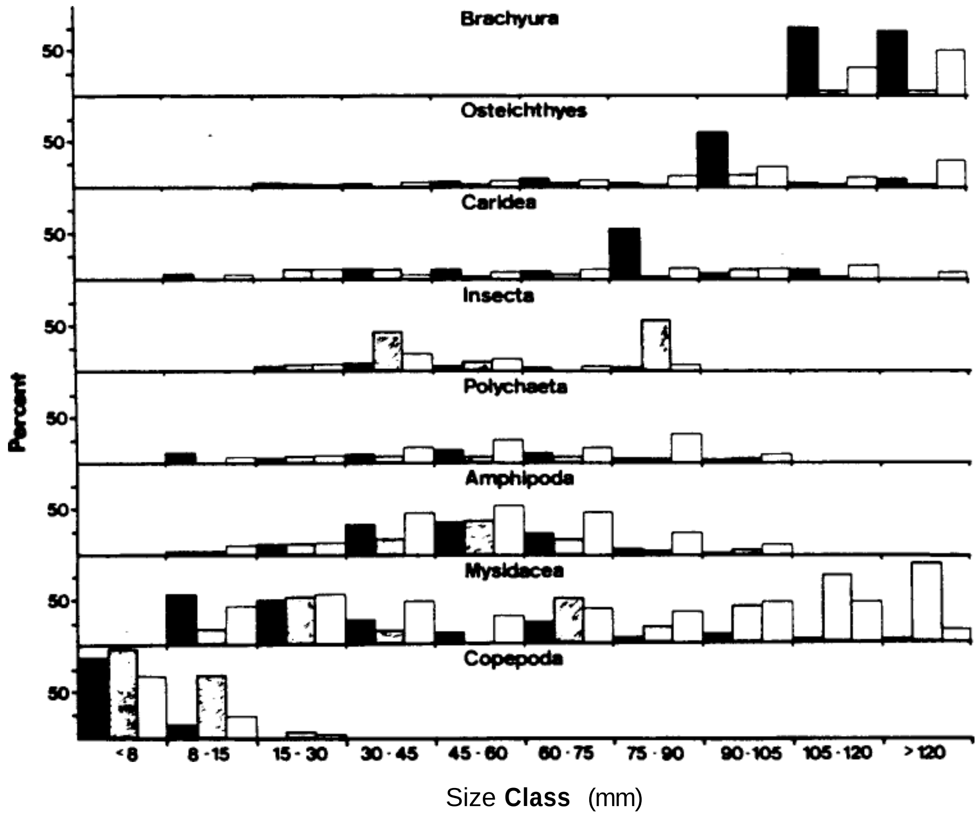

Figure 3-20. Percent volume (striped), number (stipled), and occurrence (open) of major prey items from 15-mm size classes of *S. ocellatus* collected in Tampa Bay. Source: Peters and McMichael (1987).

areas. Trace metal poisoning was suggested as a possible cause of mortality in adult red drum in the Indian River during 1980. High levels of copper, zinc, arsenic, chromium, cadmium, and mercury were found in the liver and/or gills (Cardeilnac et al. 1981; cited from Mercer 1984). The mean range of PCBs in five red drum (no size given) from Texas was 0.03 ppm (range = 0.02 - 0.04 ppm). This level was far below existing (5 ppm) or proposed (2 ppm) maximum permissible levels in food fish (Gadbois and Maney 1983; cited from Mercer 1984). While it is difficult to relate observations directly to the Tampa Bay estuary, investigations have reported variable levels of many trace metals and PCB's in sediments within the Tampa Bay estuary (Long et al. 1991). Unfortunately, no studies were available to assess contaminant concentrations in red drum in this estuary. FDNR-FMRI is currently investigating the incidence of mercury and other trace metal concentrations in fish tissues of numerous fish species within Tampa Bay (McMichael, pers. comm. 1992).

3.6.5 ENVIRONMENTAL REQUIREMENTS

Red drum are dependent on estuaries for at least their first few years of life. The Tampa Bay estuary provides many of the water quality and structural habitats required by red drum throughout their life history. The interaction of temperature and salinity parameters are important to the survival and growth of egg, larval, juvenile and adult red drum (Holt et al. 1981; Reagan 1985; Neill 1990). Specific structural habitats are also required by larval and juvenile red drum within the Tampa Bay estuary.

3.6.5.1 Salinity

Red drum juveniles and adults are euryhaline and are found naturally in fresh water, brackish water estuaries and marine waters (Simmons and Breuer 1962). They have been collected at salinities of 0 to 50 ppt in Texas estuaries (Gunter and Hall 1962).

Salinity may have an effect on egg hatching success. Red drum eggs float at salinities of 25 ppt and higher, but sink when salinities drop below 20 ppt. Eggs that sink to the bottom may become covered with silt and die (Holt et al. 1981).

In Tampa Bay, red drum larvae occur in the more polyhaline regions of the estuary. Surface water salinities where larval red drum were collected in Tampa Bay ranged from 16 to 34 ppt (Peters and McMichael 1987). Similar salinities were reported for larval red drum collected in Florida Bay (8 to 35 ppt; Rutherford et al. 1986). In the Little Manatee River area, red drum larvae (flexion and post-flexion) were most common in the more saline bay waters. With increased development, they began to enter the more freshwater portions of the tidal river (Peebles et al. 1992) (Fig. 3-21). This agrees with research conducted by Crocker et al. (1981) who evaluated survival and growth of juvenile red drum in fresh and saltwater. These authors reported that tolerance to freshwater was size dependent. They found survival rates of 5% in larvae (6.2 mm SL), 70% in postlarvae (16.2-19.7 mm SL) and 95% in juveniles subjected to freshwater for 96 hours. Survival in control salinities of 10 ppt was 90% or greater for all length groups.

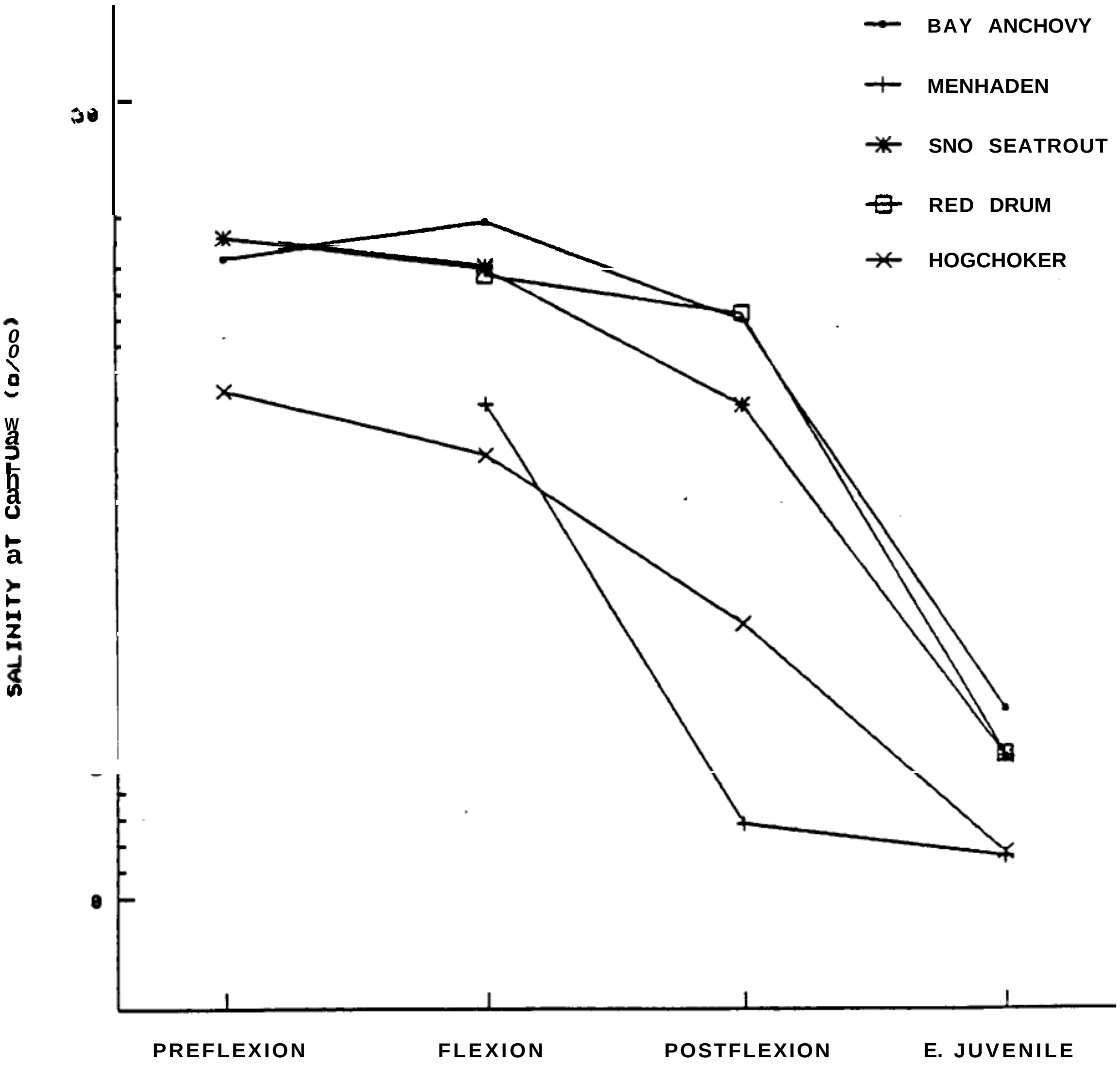

Figure 3-21. Examples of decrease in mean salinity at capture with progressing development. Source: Peebles et al. (1992).

Juvenile red drum are euryhaline and were collected in Tampa Bay waters with salinities of 0 to 37 ppt during 1987-1991 (FIMP, unpublished data). Similar salinity ranges (2 to 40 ppt) were reported in Florida Bay (Rutherford et al. 1986). Juvenile red drum prefer oligohaline (0.5-5 ppt and mesohaline portions of 5-18 ppt) tidal tributaries within the Tampa Bay estuary. In the Little Manatee River, juveniles preferred oligohaline (0.5-5 ppt) salinity regions (Fig. 3-22) (Haddad et al. 1992). Red drum were nearly always most abundant at a station located upriver at river kilometer 5.8 with catch rates generally declining both up- and down-stream from this site (Fig. 3-23). During 1990, recruitment to the preferred station was lower than previous years. Coincidentally, this represented the only fall period when the favored site was within the mesohaline (5-18 ppt) rather than oligohaline (0.5-5 ppt) zone and represented 'the winter period with highest average salinities at this site. There appeared to be little doubt that red drum juveniles entering the Little Manatee River preferentially inhabited the middle and lower portion of the salinity gradient (Haddad et al. 1992).

In the Manatee River, juvenile red drum were collected at stations with salinities ranging from 0 to 21 ppt. However, red drum were significantly more numerous in collections at stations with salinities ranging from 16 to 20 ppt. Over half (n=388 total) of the red drum were caught in this salinity range although only 25% of the stations fell within this range (Edwards 1990). These salinity preferences are higher than those observed in the Little Manatee River which suggests that salinity may not be the only factor affecting the distribution of juvenile red drum in tidal tributaries. For example, variable salinity within tidal tributaries could affect distribution of prey items, and therefore could indirectly effect red drum distribution (Peebles, pers. comm. 1992).

The interaction of salinity and water temperature is important to the life history of red drum. Optimal conditions for hatching and 24 hr survival are influenced by salinity and temperature salinity interactions, whereas temperature was a substantial factor in larval survival and growth beyond 24 hours (Holt et al. 1981). Holt et al. (1981) examined egg hatching and larval survival under laboratory conditions. Four salinities (15, 20, 25 and 30 ppt) and three temperatures (20, 25 and 30°C) were compared to identify optimal salinity and temperatures. A pattern of decline in percent egg hatch with increases in temperature at lower salinities (15 and 20 ppt) but not at higher salinities was indicative of a temperature-salinity interaction. Hatching rates were significantly higher at the two higher salinities than lower salinities. Higher temperatures and lower salinities usually decreased hatching success. The optimum combination of temperature and salinity for hatching and 24 hr. larval survival was 25°C and 30 ppt.

3.6.5.2 Temperature

Water temperature plays a major role in egg hatching time, duration of yolk sac stage and early growth rates. Duration of yolk sac stage ranges from 40 hrs at 30°C to 85 hrs at 20°C (Holt et al. 1981). Optimum growing temperature was 27°C and in temperatures of 20°C or lower, larval red drum become inactive, ceased feeding and

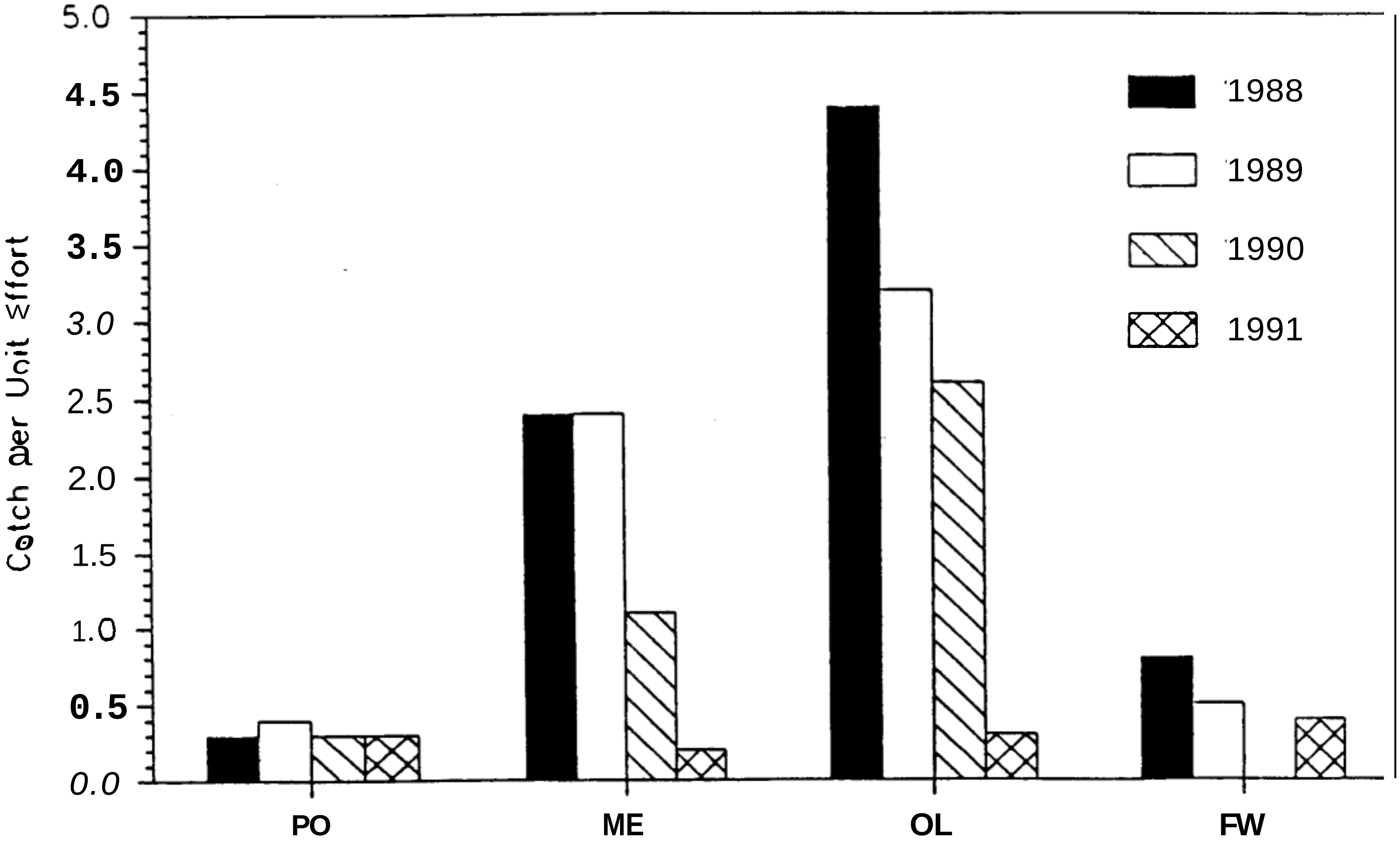

Figure 3-22. CPUE (# fish/100 m²) for juvenile red drum by surface salinity in the Little Manatee River. PO = polyhaline (> 18 ppt); ME = mesohaline (5-18 ppt); OL = oligohaline (0.5-5 ppt) and FW = freshwater (0-0.5 ppt). Source: Haddad et al. (1992).

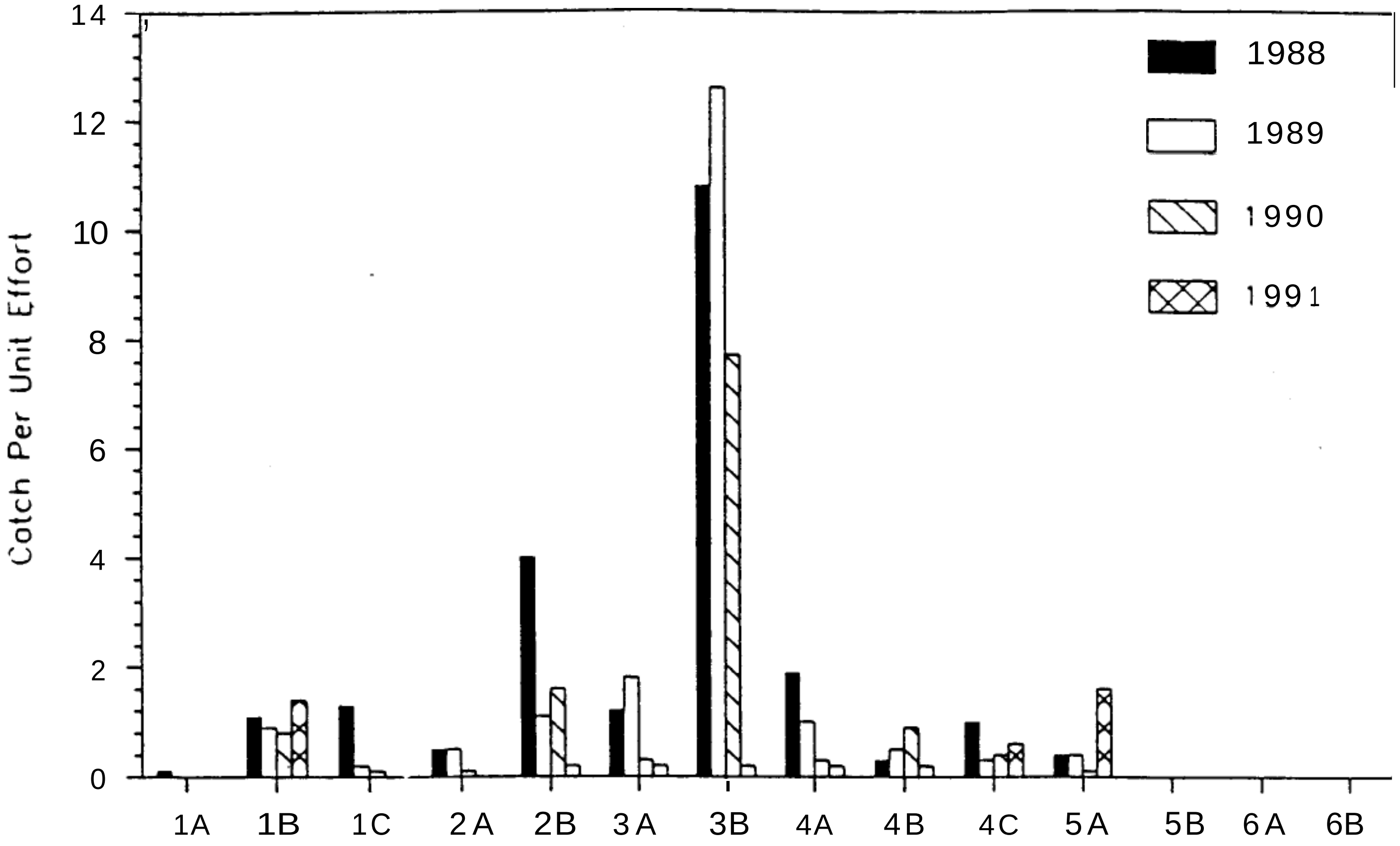

Figure 3-23. CPUE (# fish/100 m^2) for juvenile red drum by location in the Little Manatee River station 1 = rkm 0; station 2 rkm 2.5; station 3 = rkm 5.8; station 4 = rkm 11.8; station 5 = rkm 11.8; and station 6 = rkm 16.4. Source: Haddad et al. 1992.

eventually died. Growth rates were positively correlated with temperature. Fourteen day growth at 20°C was 0.1 mm/day; at 25°C was 0.2 mm/day and at 30°C was 0.3 mm/day (Holt and Arnold 1982). Lee et al. (1984) raised larval red drum at 24 and 28°C (salinity **30** ppt). Larvae grown at higher temperatures were significantly larger and heavier after 15 days. These authors suggested that the spawning success of red drum depends, to a large extent on when peak spawning occurs, and that larvae hatched in November and December will have lower survival than those produced in September and October because of lower water temperatures.

Red drum tolerate a wide range of water temperatures in the Gulf of Mexico (Perret 1971; Simmons and Breuer 1962). Larval and juvenile red drum were collected in Tampa Bay in water temperatures ranging from 12.5-32.2°C (Peters and McMichael 1987; FIMP unpublished data). Red drum are generally considered to be eurythermal; however, sudden freezing temperatures can lead to mass mortalities (Storey and Gudger 1936; Gunter 1941; Gunter and Hildebrand 1951). Buckley (1984) suggests that the rate of temperature change is more important than the lowest temperature reached. A wide temperature range of 2-33°C can be tolerated as long as the change is gradual (Gunter and Hildebrand 1951; Simmons and Breuer 1962). Gilmore et al. (1978) reported no observations of red drum mortality in the 1977 freeze which occurred in Tampa Bay. Red drum may move to deeper, more thermally protected Waters in times of extremely low water temperatures. Rinckey and Saloman (1964) report that during a hypothermal fish kill in Tampa Bay during December, 1962, no red drum were collected in areas of low water temperature (9.6°C) although they were collected from that area during the previous month.

3.6.5.3 Dissolved Oxygen

Little information is available on dissolved oxygen (D.O.) requirements of red drum. Neill (1990) reported that critical D.O. concentration for juvenile red drum is about 2 ppm in 5 to 10 ppt seawater, at 24°C. He assumed that the critical oxygen concentration would be least at optimum salinity and less at low than at higher temperatures. Low dissolved oxygen levels in Laguna Madre, Texas have caused fish kills which included red drum (Miles 1950).

3.6.5.4 Water Depth

Juvenile red drum (< 100 mm) appear to prefer shallow estuarine waters (< 2 m deep) within Tampa Bay. Large numbers of juveniles were taken in a shallow (0.25-2.0 m) man-made canal and a shallow (1-1.5m) tidal creek in the Alafia River (Peters and McMichael 1987). Juveniles preferred a shallow (0-.60 m) depositional shoreline in the Little Manatee River (Haddad et al. 1992). Juveniles have also been collected along shallow (< 2 m) areas within Tampa Bay. Juveniles reportedly leave these shallow areas for deeper bays and bayous as their size increases. Peters and McMichael (1987) report that many studies have found reduced numbers of red drum in shallow waters at sizes greater than 80 mm. As juveniles approach 200 mm during their first spring, they may remain in deep basins or bayous (> 2 m) venture into shallows (< 2 m) or congregate near passes (Pearson 1929; Miles 1950; Simmons and Breuer 1962; cited from Peters and McMichael 1987).

3.6.5.5 Structural Habitat

A wide range of nursery habitats are used by larval and juvenile red drum in the Tampa Bay estuary. Pelagic larvae were collected in the open waters of Tampa Bay with plankton nets (Peters and McMichael 1987). Larvae were also collected in shallow seagrass areas in the lower and mid-bay regions. Small juveniles (10-20 mm SL) were collected in seagrasses and along shorelines in the Tampa Bay estuary (Peters and McMichael 1987). Use of shallow seagrass habitats by post-larval and early juvenile red drum (> 7 mm) has also been reported in other estuaries (Holt et al. 1983; Rutherford et al. 1986).

Red drum appear to change habitats with growth and larger juveniles apparently migrate to lower salinity backwater areas and tidal tributaries. Springer and Woodburn (1960) collected numerous juvenile (20-126 mm) red drum in a low salinity canal with a muddy unvegetated bottom. Peters and McMichael (1987) reported that juvenile (30-60 mm) red drum were very abundant in backwater regions in the Alafia River. Juveniles (60-134 mm SL) were found mainly in backwaters, although they were less concentrated in these areas than at smaller sizes. Studies have reported that red drum were also abundant in backwater areas of the Little Manatee and Manatee Rivers (Edwards 1990; Haddad et al. 1992). Common habitat characteristics of these backwater areas included: unvegetated muddy bottoms, low energy shorelines or cove areas, and some emergent vegetation along shorelines (e.g., *Rhizophora mangle, Spartina sp., Juncus sp.).*

Structural habitats of subadult and adult red drum have not been adequately described for the Tampa Bay estuary. Peters and McMichael (1987) report that at increased sizes, juveniles and subadult become less common in the backwater regions and apparently move to deeper water portions of the estuary.

Environmental requirements of red drum are summarized in Table 3-10.

3.7 SILVER PERCH *(Bairdie//a chrysoura)*

3.7.1 INTRODUCTION

Silver perch is a common sciaenid in nearshore coastal and estuarine waters. It ranges from New York to Florida and along the Gulf of Mexico (Hoese and Moore 1977; Kleypas and Dean 1983). Springer and Woodburn (1960) reported 13 species of sciaenids

Table 3-10. General and preferred ranges and upper and lower tolerance limits for environmental requirements of red drum. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	**Lower Limit**	**Upper Limit**	**Range**	**Reference**
Temperature (°C)	**25-30 (E,L)**			**20-30 (E,L)**	**Holt et al. 1981**
				12.5-32.2 (J)	**Peters and McMichael 1987** **FIMP (unpubl. data)**
				2-33 (J)	**Gunter and Hildebrand 1951** **Simmons and Breuer 1962**
Salinity (ppt)	**39 (E,L)**			**15-30 (E,L)**	**Holt et al. 1981**
				16-34 (L)	Peters and McMichael 1987
				8-35 (L)	Rutherford et at. 1986; Haddad et al. 1992
				0-37 (J)	FIMP (unpubl. data)
				2-40 (J)	Rutherford et al. 1986
				0-21 (J)	Edwards 1990
D.O. (mg/L)		2 (J)			Neill 1990
Depth (m)	variable (E,L)				Robinson 1985; Holt et al. 1983; Peters and McMichael 1987
	0.25-2 (J)				Peters and McMichael 1987
Substrate	seagrasses (L,J)				Holt et al. 1983
	seagrasses, backwaters, muddy bottoms (L,J)				Peters and McMichaels 1987; Springer and Woodburn 1960; Edwards 1990

occur in the Tampa Bay region, with silver perch being one of the most abundant. Lewis (1989) listed this fish as one of the 10 dominant species in the Tampa Bay estuary. Silver perch are of no commercial and of minor recreational importance to the estuary. However, given their abundance in the estuary, they probably contribute significantly to the ecology of the ecosystem.

3.7.2 LIFE HISTORY

Early life history of the silver perch has been examined in the Tampa Bay estuary. Relative abundance of small larvae (< 3 mm) decreased from lower (Sunshine Skyway) to mid (Cockroach Bay area) and upper bay (Alafia River area) collections, suggesting that spawning occurred in the lower bay or nearshore Gulf waters (Peters and McMichael unpublished manuscript).

Silver perch appear to spawn year round in south Florida estuaries (Tabb and Manning 1961, Jannke 1971). Peters and McMichael (unpublished manuscript) reported that the presence of larval silver perch during most months suggested that this species also spawns year round in the Tampa Bay estuary. Larval abundance peaked in April and May, although secondary peaks were noted in August, September, and January. Peebles (in prep.) collected silver perch larvae near the Little Manatee river from April through September.

Silver perch larvae exhibited two trends in vertical distribution (Peters and McMichael, unpublished manuscript). Larvae were negatively phototaxic (moving closer to surface water during periods of darkness) and larger larvae showed a preference for bottom waters.

Small (< 30 mm) juvenile silver perch are present during most months in the Tampa Bay estuary, supporting the theory of year-round spawning (Fig. 3-24; Haddad et al. 1992; Table 3-11; FIMP unpublished data). Juvenile silver perch recruited primarily to structured habitats such as seagrass beds, rocks, and seawalls at lengths of 8-25 mm SL. Juveniles greater than 25 mm SL were abundant in seine collections from seagrass beds and tidal creeks (Peters and McMichael, unpublished manuscript). Large juveniles and adults may migrate to deeper bay and offshore waters (5-7 m), especially during winter months (Moe and Martin 1965). All lifestages of silver perch appear to prefer waters of mid to high salinities although they are found in lesser numbers in lower salinity backwater areas of the Tampa Bay estuary.

Juvenile growth rates derived from modal increases in length frequency distributions were estimated at 7-20 mm SL/month (Peters and McMichael, unpublished manuscript). These authors reported that these rates were slightly slower than other studies in the Gulf of Mexico and Atlantic Ocean which reported growth at 10-23 mm SL/month (Clark 1971) or 17-28 mm SL/month (DeSylva et al. 1962; Chao and Musick 1977). Moe and Martin (1965) reported that growth of the first year class appears to

Table 3-11. Density (# fish/m^2), mean length (mm SL), and range of lengths of juvenile silver perch collected at some fixed stations as part of Florida's fisheries independent monitoring program (FIMP 1990)

	Density	Mean	Min	Max
Station 6 (4th Street Causeway)				
Jun	.20	41	14	49
Jul	.16	48	32	79
Aug	3.03	47	37	78
Sep	.08	56	38	89
Jun	1.1	50	37	71
Jul	.37	52	16	86
Aug	.09	37	10	97
Sep	.06	19	14	29
Station 8 (Big Bayou)				
May	.06	32	26	48
Jun	.83	49	14	72
Jul	1.23	55	32	75
Aug	.57	57	25	84
Sep	.55	66	18	103
Oct	.19	84	26	122
Station 11 (Simmons Park)				
Apr	.01	12	8	15
May	.34	15	7	32
Jun	1.16	35	26	55
Jul	4.57	49	28	64
Aug	.49	26	12	63
Sep	.57	37	13	92
Oct	.12	51	38	85

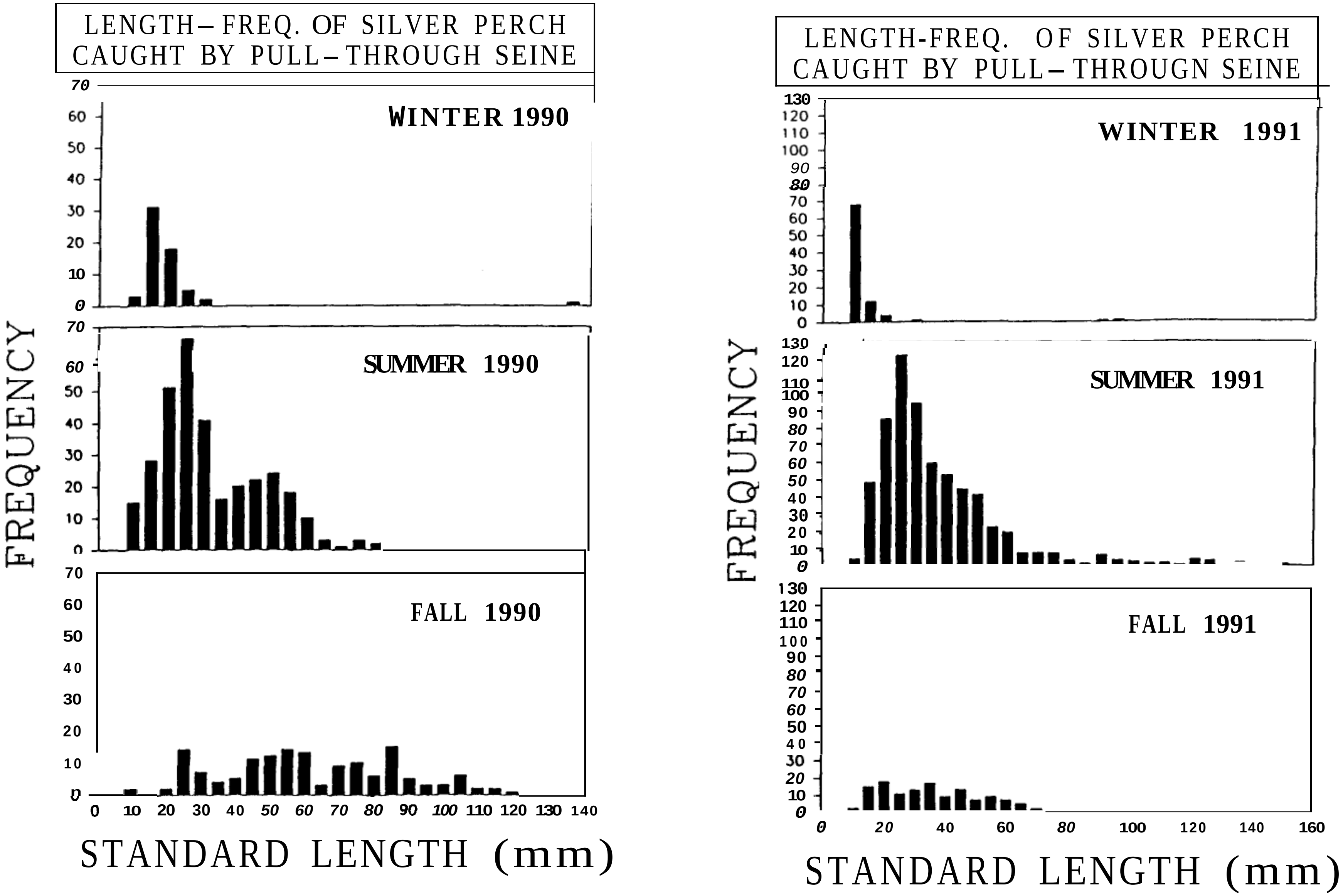

Figure 3-24. Length-frequencies of silver perch caught in shallow seagrass beds outside the Little Manatee River during 1990 and 1991 (Haddad et al. 1992)

increase with increasing water temperature, and that maturity may be achieved during the first year.

3.7.3 ECOLOGICAL ROLE

Silver perch are primarily benthic carnivores with crustaceans being the most dominant food source. Hildebrand and Cable (1930) reported feeding ecology of silver perch near Beauford, NC. Juveniles 7-20 TL mm fed primarily on copepods with some ostracods, amphipods, cladocerans, and a few mysids and polychaetes. Fish 25-50 mm TL fed primarily on mysids, small shrimp and crabs and less of copepods, ostracods, isopods, and polychaetes. Silver perch 50-80 TL mm fed predominately on mysids, shrimp, gammarid amphipods, and polychaetes. Adult diet included that of the previous size class as well as small fishes such as Anchoa spp. Similar feeding habits have been reported along the west coast of Florida. Near Cedar Key, FL, silver perch 25-99 mm long fed primarily on shrimp, copepods and amphipods with smaller amounts of mollusks, polychaetes, fishes and crustaceans. Fish 100-130 mm fed mainly on shrimp and crustaceans, with amphipods, crabs and fish being less important (Reid 1954). Carr and Adams (1973) provided a detailed analysis of stomach contents of approximately 800 juvenile silver perch collected near Crystal River, FL. Nineteen size classes between **6** and 160 mm SL (5 mm increments) were examined (Fig. 3-25). The youngest stages fed exclusively on planktonic copepods. Copepods decreased in importance in larger specimens. Silver perch 16-40 mm showed an increased consumption of mysids and small shrimp. Food items of occasional significance in larger specimens (91-100 mm) included small fish (pinfish, gobiids, and silver perch) amphipods, and polychaetes. Springer and Woodburn (1960) reported general information on silver perch feeding ecology in the Tampa Bay estuary. Crustaceans of various groups (amphipods, copepods, mysids, gammarids, caridean and penaeid shrimp, crab larvae) and to a lesser extent fishes, polychaetes and insect larvae, formed the diet. A quantitative study of juvenile silver perch feeding habits in Tampa Bay suggest that a large proportion of the diet (by volume and number) is comprised of calanoid copepods, mysids and gammarid amphipods (see Peebles 1992).

3.7.3.1 Predators

Little information is available to document predators of silver perch. Silver perch have been reported as prey items of juvenile spotted seatrout in Tampa Bay (McMichael and Peters 1989; Peebles 1992). Considering the abundance of this species in the Tampa Bay estuary, it is likely that they are prey to numerous species of piscivorous fish.

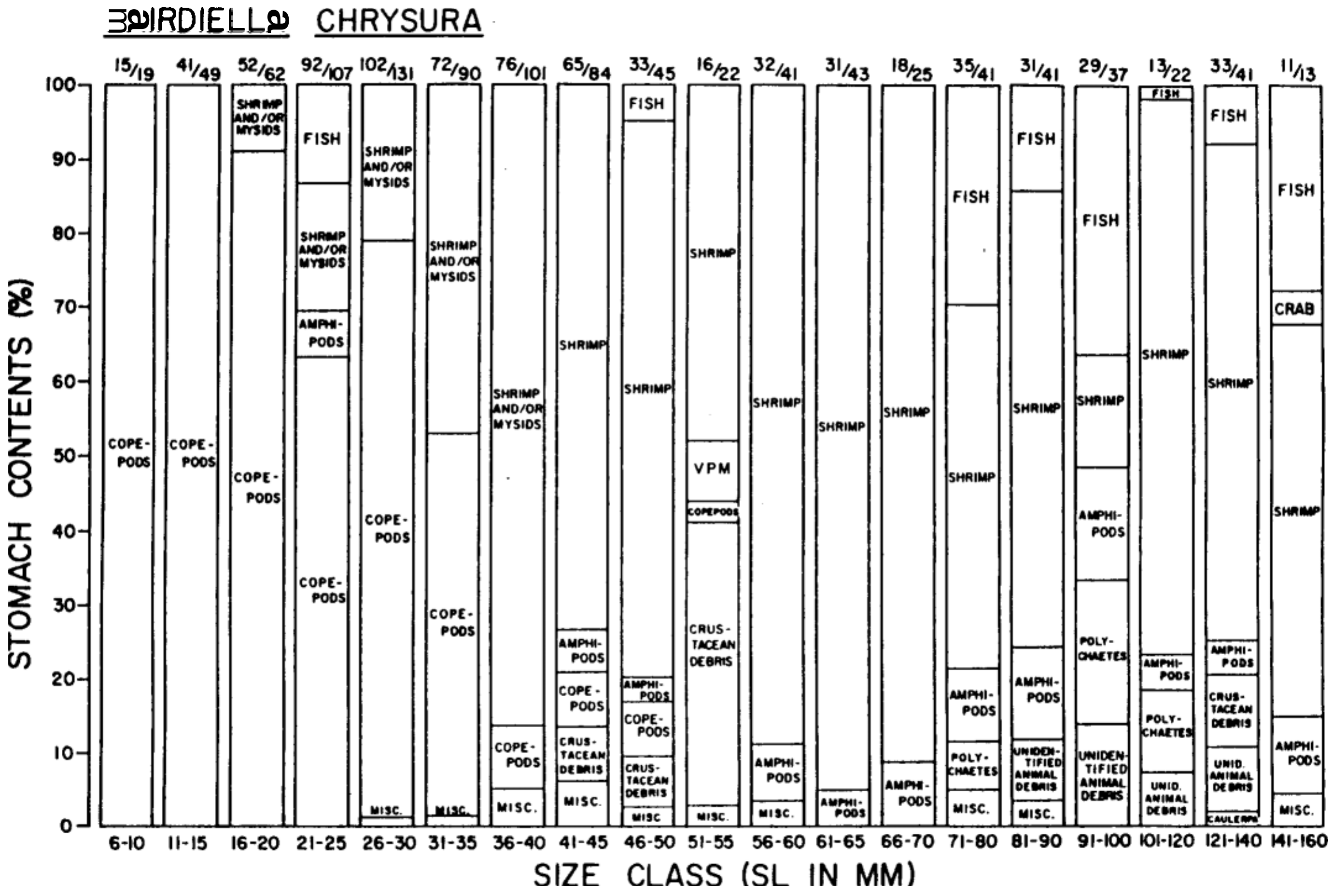

Figure 3-25. Stomach contents of juveniles of *Bairdiella chrysura*. "VPM" = vascular plant material. Source: Carr and Adams (1973)

3.7.4 ENVIRONMENTAL REQUIREMENTS

It appears that a variety of water quality and structural habitat parameters may influence the distribution and abundance of silver perch in the Tampa Bay estuary. Specific environmental requirements will be discussed below.

3.7.4.1 Salinity

Silver perch are a euryhaline species which appear to have a preference for mid to high salinities. Larvae have been collected in salinities ranging from less than 1 ppt to 32 ppt (Peters and McMichael, unpublished manuscript). However, most were taken in salinities greater than 10 ppt. Jannke (1971) reported that larval silver perch in Florida Bay also preferred these higher salinity waters.

Juvenile silver perch in Tampa Bay were collected in salinities ranging from 0 to 35 ppt, although most were in salinities over 20 ppt (Springer and Woodburn 1960; Peters and McMichael, unpublished manuscript). Juveniles are often a dominant species throughout the higher salinity portions of the estuary, but are less abundant in lower salinity backwaters and tributaries (FIMP 1990).

In the Little Manatee River region, juvenile silver perch had their peak abundances located in meso- and polyhaline salinities nearer the mouth of the river generally between rkm 0-4 (Figs. 3-26 and 3-27) (Haddad et al. 1992). However, a few individuals were collected in oligohaline and freshwaters as far upstream as rkm 16.4 during some years. Adult silver perch were commonly collected in shallow coastal areas just outside the Tampa Bay mouth, where salinities are typically marine (> 30 ppt) (Moe and Martin 1965). In general, it appears that although silver perch is a euryhaline species, they prefer areas in the Tampa Bay estuary characterized by moderate to high salinities.

3.7.4.2 Temperature

Silver perch is a eurythermal species which have been collected in a wide range of water temperatures (10 - 32.5°C) in the Tampa Bay estuary (Springer and Woodburn 1960). They are tolerant of typical warm water conditions within the estuary; however, extreme low water temperatures caused by seasonal cold fronts may result in mortalities. Chung (1977) determined upper temperature tolerance limits for silver perch collected in Galveston Bay, Texas. Fish 20 - 200 mm had three hour LD_{50}s at 34 - 37°C and 30 min. LD_{100}s at 37 - 40°C. Silver perch have been collected in Tampa Bay waters in temperatures as low as 10°C, although some cold water induced mortalities were observed in waters of 13°C during December 1957 (Springer and Woodburn 19601. No mortalities were reported in cold water fish kills reported in Tampa Bay during 1964 or 1977 when water temperatures dropped as low as 9.6°C in some areas (Rinckey and Saloman 1964; Gilmore et al. 1978).

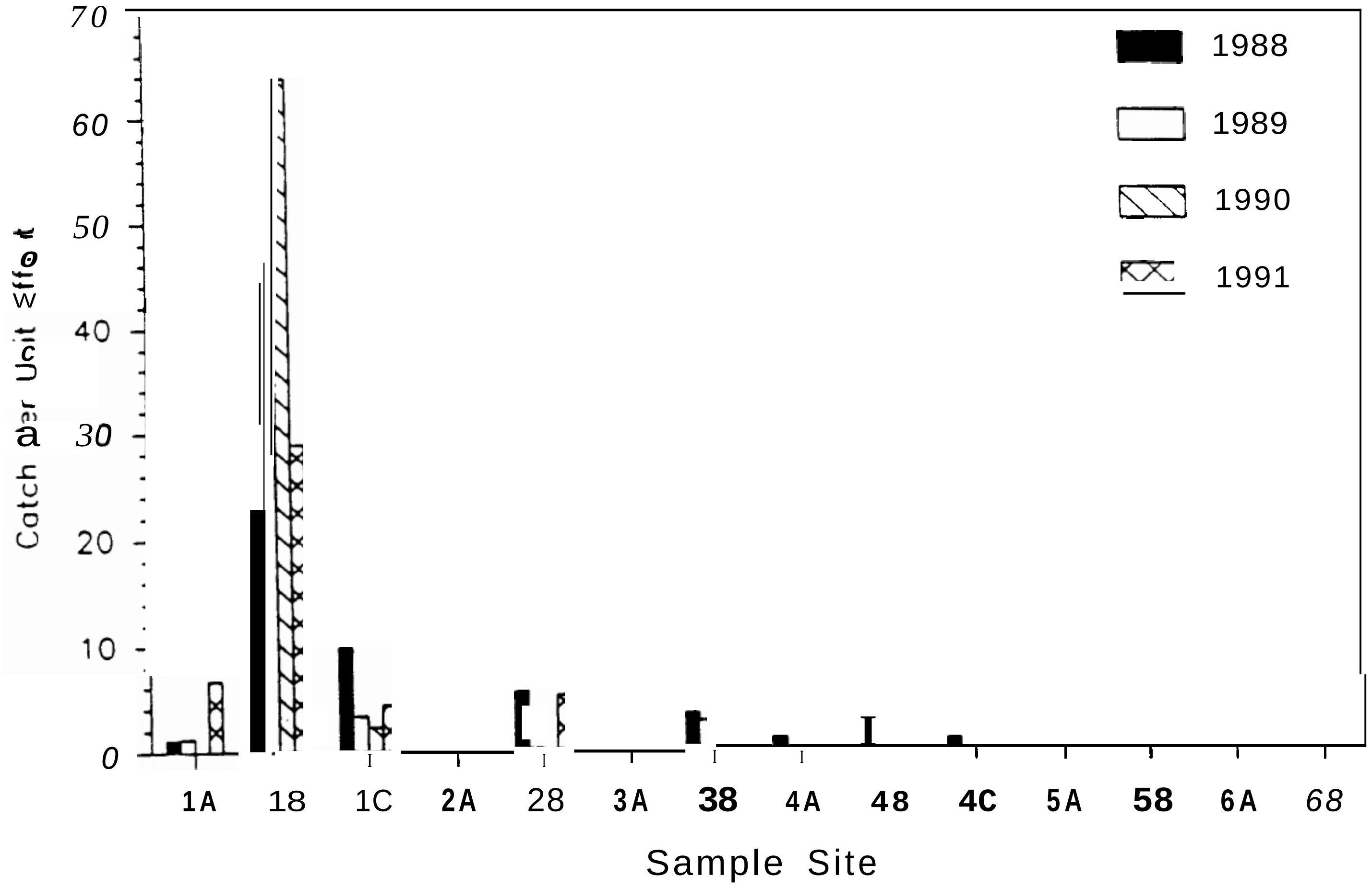

Figure 3-26. Catch per unit effort (# fish/100 m^2) for juvenile silver perch by station location in the Little Manatee River. Station 1 = rkm 0, Station 2 = rkm 2.5, Station 3 = rkm 5.8, Station 4 = rkm 11.8, Station 5 = rkm 15.5, Station 6 = rkm 16.4. Source: Haddad et al. 1992.

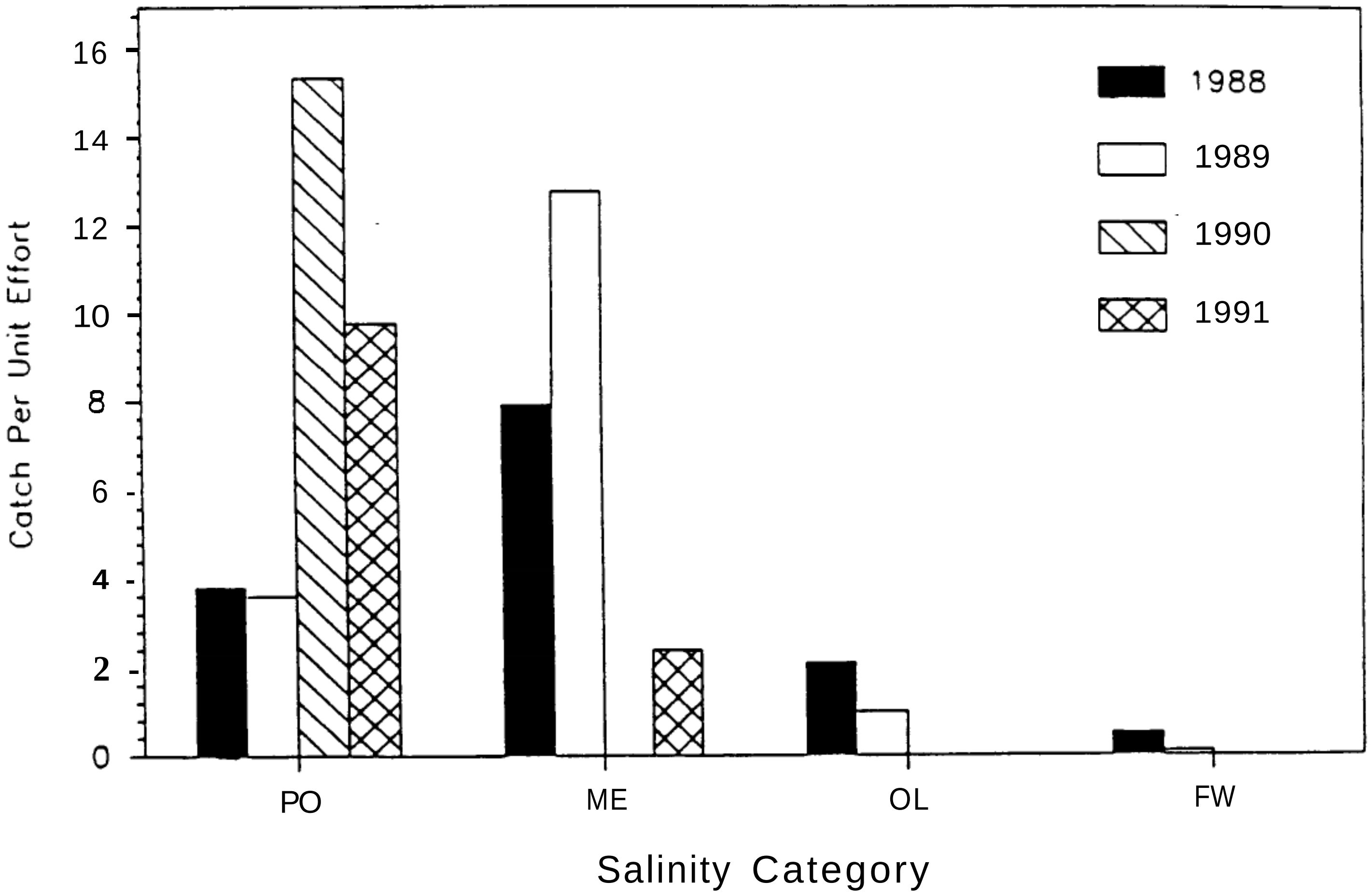

Figure 3-27. Catch per unit effort (# fish/100 m^2) for juvenile silver perch by salinity category. PO = polyhaline (> 18 ppt), ME = mesohaline (5-18 ppt), OL = oligohaline (0.5-5 ppt), and FW = freshwater (0-0.5 ppt). Source: Haddad et al. (1992).

Silver perch show distinct patterns in seasonal abundance which suggested that they may abandon shallow areas for more thermally protected deeper waters during winter months. Juveniles were rarely collected in shallow waters near the mouth of the Little Manatee River during winter months (Jan-Apr) although they were common there throughout the remainder the year (Fig. 3-28; Haddad et al. 1992). **No** juvenile silver perch were collected in shallow water drop net collections taken throughout Tampa Bay during November months (1987-1990), although they were common in May and August collections (Fonseca, unpublished data 1992). Moe and Martin (1965) reported larger catches of silver perch in nearshore coastal waters outside Tampa Bay during winter months versus spring and summer. These authors speculated that decreased abundance of this species offshore during warmer months was due to spawning migrations to inshore waters. Silver perch have also exhibited distinct seasonal trends in other estuaries (DeSylva et al. 1962; Storer 1983; Sogard et al. 1989).

3.7.4.3 Dissolved Oxygen

Very little information was available to determine the dissolved oxygen requirements of silver perch in Tampa Bay. Silver perch were collected in waters with D.O.'s ranging from 1.3 to 15.2 ppm (FIMP, unpublished data, 1987-1991). No information on lethal levels of D.O. was available.

3.7.4.4 Water Depth

Silver perch prefer shallow (< 2 m) seagrass and backwater habitats in warmer months, whereas larger juveniles and adults may move to deeper (5-7 m), more thermally protected waters in winter (Moe and Martin 1965; FIMP 1990; Haddad et al. 1992).

3.7.4.5 Structural Habitat

Silver perch appear to transform from a planktonic to a demersal existence between 8 and 25 mm SL, when they tend to associate with structural or live bottom habitats such as rocks, piers, seawalls and seagrass beds. Juvenile silver perch were collected in many habitats but were most abundant around structured habitat at small sizes and more limited to seagrass beds at larger sizes (Peters and McMichael, unpublished manuscript). Juveniles were more abundant in collections from seagrass than in samples from rivers and bayous lacking seagrass.

Silver perch occupy areas in Tampa Bay characterized by a wide variety of submerged aquatic vegetation including: *Thalassia testudinum, Syringodium filiforme, Halodule wrightii, Ruppia sp.* and *Caulerpa sp.* (Fonseca, unpublished data 1992). This species is often listed as numerically dominant in many seagrass habitats (FIMP 1989, 1990). Haddad et al. (1990) reported high (0.35/m^2) densities of silver perch near the mouth of the Little Manatee River in areas of moderate to dense seagrass *(Halodule sp.)*

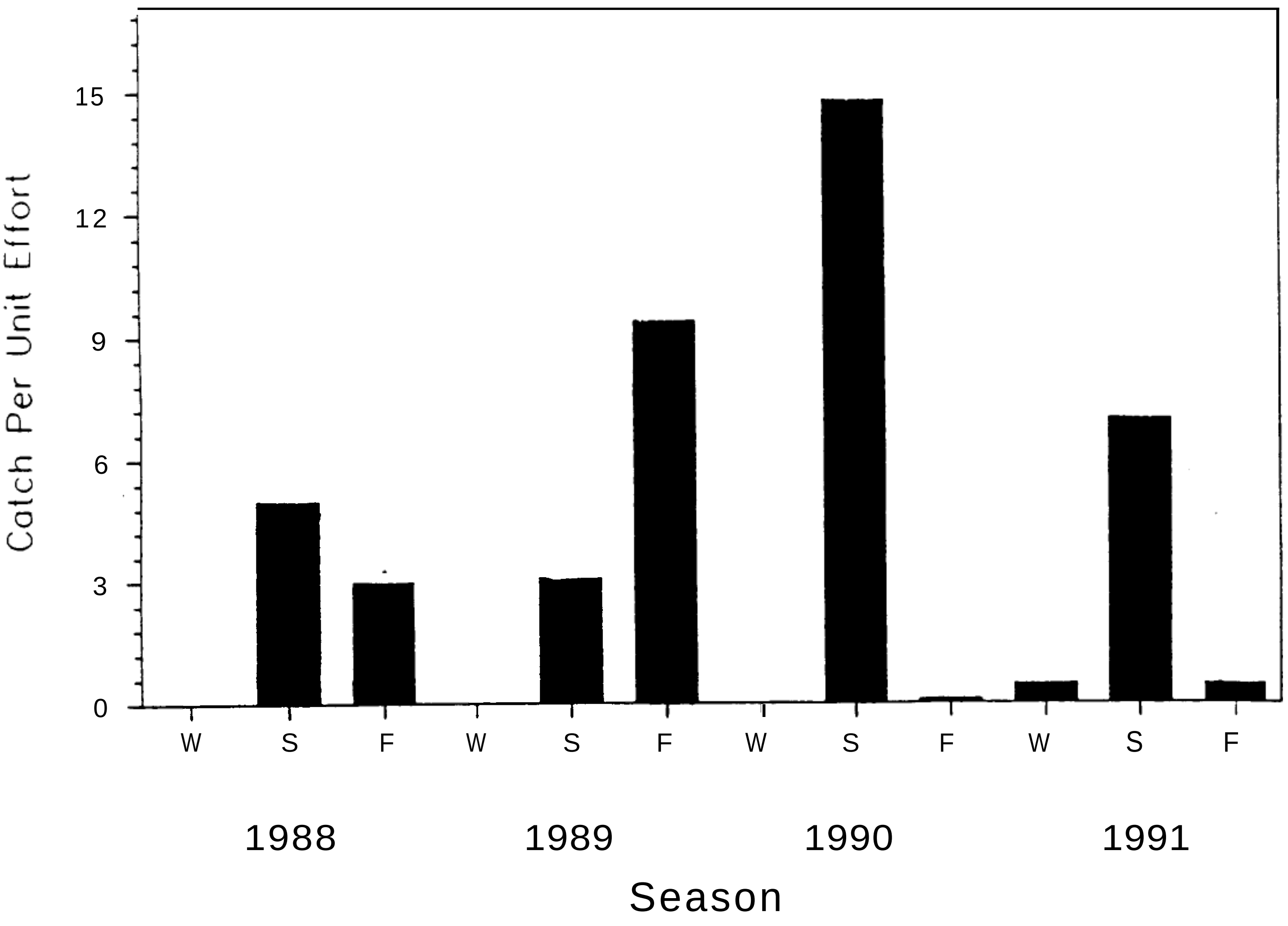

Figure 3-28. Catch per unit effort (# fish/100 m^2) of juvenile silver perch by season in the Little Manatee River. Source: Haddad et al. (1992).

and macroalgae. Gilmore et al. (1978) reported similar high densities (mean = 0.1 6/m^2) in seagrass habitats on the east coast of Florida. Silver perch densities were highest in moderate seagrass densities and lower but still substantial in dense seagrass. Densities in sparse and very sparse grass and over bare sand were quite low. There was no recognizable relationship between silver perch density and macroalgae density (Haddad et al. 1992).

Silver perch are much less common in shallow unvegetated and backwater habitats within the Tampa Bay estuary (FIMP 1989, 1990; Price and Schleuter 1985). Similar observations have been documented in Florida Bay (Thayer et al. 1987). In general, it appears that although juvenile silver perch are found in a wide variety of habitats, including backwater areas, tidal tributaries, and over bare sediment, they prefer shallow vegetated seagrass regions. Adults are commonly collected in sandy unvegetated nearshore habitats outside the mouth of the Tampa Bay estuary during winter months, which suggests some degree of habitat partitioning with growth (Moe and Martin 1965). However, this may also be a result of avoidance of colder winter water temperatures in the estuary.

Environmental requirements of silver perch are summaried in Table 3-12.

3.8 SPOT *(Leiostomus xanthurus)*

3.8.1 INTRODUCTION

Spot is an estuarine dependent sciaenid found in near and offshore waters depending upon life stage. Spot are abundant along the Gulf of Mexico and Atlantic coasts of the United States from Texas to New York, and have been reported as far north as the Gulf of Maine (Bigelow and Schroeder 1953). Spot inhabit a wide variety of habitats, including estuarine, marine and freshwater areas (Springer and Woodburn 1960; Homer and Mihursky 1991).

Spot is one of the most common fish species found in the Tampa Bay estuary. Spot supports a small but notable commercial fishery. Commercial harvest in Tampa Bay has declined from 86,700 pounds in 1986 to 14,830 pounds in 1990, although a reduction in fishing effort may have contributed to some of this decline (Table 3-13). Spot is one of approximately 79 fish species which use Tampa Bay as a nursery habitat (Springer and Woodburn 1960; Comp 1985) and is listed among the top 10 dominant species found throughout the bay (Comp 1985; Lewis and Estevez 1988). This species is considered a major regulator of benthic invertebrate species and is important in the structure and function of estuarine ecosystems (Phillips et al. 1989; Homer and Mihursky 1991).

Table 3-12. General and preferred ranges and upper and lower tolerance limits for environmental requirements of silver perch.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)		13°		10-32.5°C	Springer and Woodburn 1960
			34-37 (3-hr LD_{50})		Chung 1977
			37-40 (30 min LD_{100})		Chung 1977
Salinity (ppt)				1-32 (L)	Peters and McMichael unpubl. manuscript
	>10 (L)				Peters and McMichael unpubl. manuscript
	>20 (J)				Springer and Woodburn 1960
Dissolved Oxygen (mg/l)				1.3-15.2	FIMP unpubl. data
Depth (m)	<2 (J)				FIMP 1989, 1990
				to 5-10	Moe and Martin 1965
Substrate	seagrass, rocks piers, seawalls (L)				Peters and McMichael unpubl. manuscript
	seagrass (J)				Peters and McMichael unpubl. manusript
				sandy, unvegetated	Moe and Martin 1965

Table 3-13. Tampa Bay; landings and effort for Spot by year 1986-90

Year	Spot	
	Pounds	Fishing Trips
86	86742	936
87	42081	1349
88	59765	1726
89	24784	1564
90	14833	1267

Source: Brown, pers. comm. 1992.

3.8.2 LIFE HISTORY

In the northern Gulf of Mexico, spot begins its life cycle by spawning along the inner continental shelf (Dawson 1958; Nelson 1969; Durako et al. 1988; Peebles et al. 1992). Eggs are released into the water column and the density of spawning individuals determines the success rate of fertilization (Dawson 1958; Nelson 1969; Joseph 1972). In the Tampa Bay region spawning occurs offshore from October through March or April (Peters and McMichael, pers. comm. 1992). The average mature female spot (200 mm SL) releases 70,000 to 130,000 eggs per spawning period (Homer and Mihursky 1991). Fertilized spot eggs are pelagic. After hatching, larval size ranges from 1.6 to 1.7 mm (Powell and Gordy 1980). Larval spot grow rapidly in warmer offshore waters coincident with winter plankton peaks, but growth slows as they enter cooler estuarine waters (Turner 1981; Warlen and Chester 1985, cited from Homer and Mihursky 1991). Powell and Gordy (1980) have indicated that warmer temperatures in the Gulf of Mexico may actually speed fin development in spot. They appear to begin this process at a much smaller size than fish spawned in cooler waters.

As with most estuarine dependent species spawned offshore, spot are either passively transported into estuaries or actively follow salinity gradients (Comp 1985). Larval spot may use tidal circulation patterns to aid their transport into Tampa Bay. By remaining in "near-bottom" waters during all tidal phases, larval fish may have more success moving into and remaining in estuarine nursery areas (Robison 1985). Winter cold fronts, with their resultant winds, may also influence the transport of fishes through inlets and mouths of estuaries (Lyczkowski et al. 1990).

Young spot spend most of their larval stage moving from offshore spawning grounds to estuarine waters. They first appear in Tampa Bay in late November through early April with peak recruitment occurring in February and March (Peters and McMichael, unpublished manuscript). Comp (1985) estimated size at recruitment into Tampa Bay to range from 12 to 18 mm SL; however, Peters and McMichael (unpublished manuscript) suggest that spot may enter the bay at sizes less than 8 mm SL. Spot appear to have multiple spawning periods resulting in overlapping length frequencies of different cohorts of a single year class. This overlap makes growth estimates of these young fish difficult to assess (Peters and McMichael, unpublished manuscript).

In the Little Manatee River tributary, peak ingression of spot occur during the postlarval stage (Peebles et al. 1992) during the months of December through April (Peebles, in prep.). Peters and McMichael (unpublished manuscript) observed a pattern of recruitment along sandy shorelines of the bay and in seagrass beds for larval spot less than 20 mm SL. These protected regions prove extremely beneficial to the young spot and they grow rapidly through the spring and summer months.

From the protected tidal rivers and shallow shoreline areas, juvenile spot migrate towards deeper waters of central Tampa Bay in late summer and early fall (Peebles et al. 1992). It has been suggested that when spot reach approximately 80 mm, they seek out the higher salinity waters in the main bay (Comp 1985; Peebles et al. 1992). Seasonal temperature declines may prompt movement from shallow nursery areas such as seagrass beds and tidal rivers to offshore areas in the Gulf of Mexico (Sheridan 1979). Juvenile spot move into deeper bay waters or offshore after approximately 8-9 months in Tampa Bay to eventually mature and spawn in the Gulf of Mexico (Parker 1971). Dawson (1958) believed that the young fish remain in inshore nursery areas with local changes in distribution, until the end of their second summer, after which they begin a gradual movement back to the sea to spawn.

Peters and McMichael (unpublished manuscript) report problems associated with estimating early growth rates of spot. Their protracted spawning season results in the presence of several cohorts in a given year class, making growth estimates difficult. Also, as spot grow, the larger individuals leave the shallow nursery areas and move to deeper waters, making it more difficult to estimate sizes of these larger fish which are no longer readily sampled.

Age and growth rates of spot have been reported in some estuaries. They reach a length of 110 mm SL by the end of their first year in the mid-Atlantic area (Joseph 1972). Homer and Mihursky (1991) cited reports that suggest spot reach sexual maturity between their second and third year, with minimum sizes ranging from 135-180 mm SL in the Chesapeake Bay area. Some age four individuals have been reported with lengths ranging from 234-290 mm (Stickney and Cueno 1982). In Texas estuaries, Parker (1971) reported that spot had ripe or developing gonads at a size range of 177-214 mm TL.

Principal causes of juvenile mortality include predation and low winter temperatures associated with early recruitment (Homer and Mihursky 1991). In some areas, estimates of annual mortality for adult spot populations can reach 80% (Pacheco 1962b; cited in Homer and Mihursky 1991).

3.8.3 ECOLOGICAL ROLE

Spot are considered both opportunistic generalists and selective predators (Ellis and Coull 1989). Larval spot (1-10 mm) feed on ptoropods, pelycypods, copepod eggs, nauplii, copepodites, and small adult copepods (Govoni et al. 1983). Larval feeding is apparently influenced by visibility and size and motility of prey (Govoni et al. 1985; Govoni and Chester 1990). Post larval spot (to 23 mm SL) are visual planktivorous feeders preying primarily of pelagic calanoid copepods (Kjelson et al. 1975; Hodson et al. 1981 a).

As is characteristic of most fish, ontogenetic changes in feeding habits occur with growth and may be timed with the availability of specific food types (Kjelson et al. 1975; Peters and Kjelson 1975; Livingston 1982; Lewis and Estevez 1988). For spot, this change in diet occurs after recruitment into estuaries. At approximately 20 mm SL, spot feeding becomes more benthically oriented (Hodson et al. 1981a; Livingston 1982). Sheridan (1979) reported that juvenile spot (20-29 mm SL) consume more insect larvae and polychaetes than copepods. Hodson et al. (1981a) reported microzooplankton (primarily harpacticoid copepods) and benthos (primarily *L eptochelia rapax,* nematodes and polychaetes) were dominant food items identified in post larval and juvenile stomachs. Juveniles less than 50 mm SL feed almost exclusively on meiofauna, while spot over 50 mm SL consume macrobenthos (Chestnut 1983).

O'Neil and Weinstein (1987) suggested that spot feed opportunistically on resources present in tidal rivers and shoals. Generally spot fall into two feeding patterns. In shallow, low salinity areas they feed mainly on insect larvae, small bivalves and detritus, where as in the deeper, higher salinity sites, spot forage on polychaetes and copepods (Sheridan 1979). Dietary differences recorded at these study locations are probably due to the presence of different prey types (Currin et al. 1984; Phillips et al. 1989). Polychaetes, sand, harpacticoid copepods, and gammarid amphipods made up a large proportion of the food items (by volume) of spot in Tampa Bay ranging from 15-105 mm SL. Harpacticoid copepods, nematodes, calanoid copepods and polychaetes made up at least 90% of the food items by number (sand excluded) (see Peebles 1992).

Juvenile spot exhibited a tidally driven feeding pattern, moving into the marsh to feed at high tides (Feller et al. 1990; Archambault and Feller 1991). Juveniles had full stomachs at high tide and were least full at the end of low tide. Juveniles feed on susceptible prey within the top 2-3 mm of sediment (Ellis and Coull 1989).

Adult spot are obligate bottom feeders which feed on benthic fauna by scooping and straining prey organisms from sediments and spitting out unwanted material (Roelofs 1954; Chao and Musick 1977). Several studies have suggested that spot have a major regulating effect on benthic invertebrate communities (Homer et al. 1980; Homer and Mihursky 1991) by reducing densities and species richness (Virnstein 1977). Declines in meiofaunal abundance have been recorded during the period of influx of juvenile spot into estuarine areas (Bozeman and Dean 1980; cited from Smith and Coull 1987; Coull 1985). Laboratory studies have demonstrated that spot are extremely effective in removing meiobenthic taxa from muddy substrate, consuming seven of nine available taxa. The average depth of the forage pit which spot create is 2.2 mm, therefore, taxa located within this depth are vulnerable to predation (Ellis and Coull 1989).

Some studies have indicated that spot will forage regardless of substrate type (Ross 1980; Gerry 1981; Cowan and Birdsong 1985). However, evidence suggests that spot prefer muddy versus sandy substrate. A limiting factor appears to be their ability to sieve the coarser sediment through their gill rakers (Smith and Coull 1987).

3.8.3.1 Predators

Spot larvae (8-16 mm SL) have been reported as prey for silversides and killifish (*Fundulus* spp.) (Weinstein and Walters 1981). Large piscivorous fish such as spotted seatrout and striped bass, bluefish, and weakfish are predators of juvenile and adult spot (Pearson 1929; Hollis 1952; Dawson 1958; Manooch 1972; Merriner 1975; Thayer et al. 1976). Predation in higher salinity waters has been suggested as a limiting factor in juvenile spot production (Currin et al. 1984). The abundance, distribution, small size and relatively high rate of growth of spot suggest that this species is of intrinsic importance in the predator-prey dynamics of the Chesapeake Bay ecosystem (Homer and Mihursky 1991). Although the ecological role of this species has not been well documented in the Tampa Bay estuary, this species may be similarly important in this ecosystem.

3.8.3.2 Contaminants

Spot are known to be susceptible to various types of contaminants. Concentrations and effects of many inorganic and organic substances were summarized by Homer and Mihursky (1991). Some of the toxicants which effect spot have been reported in varying concentrations in the sediments in the Tampa Bay estuary (Long et al. 1991) and include cadmium, copper, DDT and metabolites, and dieldrin and mirex. The acute toxicity of these toxicants is reported in Table 3-14.

Homer and Mihursky (1991) reported that exposure to contaminated sediments is of particular concern for spot in Chesapeake Bay, because of their feeding habits and preference for areas with fine-grained sediments. Their benthic prey also can concentrate contaminants from sediment. They suggested spot could be an important species for monitoring tissue concentrations for persistent pollutants.

3.8.4 Environmental Requirements

Spot is an extremely adaptable species and is tolerant of a wide range of environmental factors. They are common inhabitants of environmentally stressed portions of the Tampa Bay estuary such as McKay Bay (Price and Schlueter 1985). They show little preference for specific habitats, with the exception of substrate (Homer and Mihursky 1991).

Estuarine habitats such as tidal rivers, marshes and seagrass beds play a critical role in the life cycle of spot (Chao and Musick 1977; Weinstein 1979). These areas serve as prime feeding grounds and also provide protection from predation (Zieman 1982; Lewis and Estevez 1988). Changes or deviations in these habitats could potentially affect spot populations (Hodson et al. 1981b).

3.8.4.1 Salinity

Spot are euryhaline and are found in fresh, brackish and marine waters. They have been reported in waters ranging from 0 to 60 ppt (Hedgepath 1967; Johnson 1978). Egg hatching and growth during early larval stages occurs in the higher salinities of the Gulf of Mexico. In laboratory studies, eggs hatched in 48 hours in salinities of 30-35 ppt (at 20°C)(Powell and Gordy 1980). Larvae are capable of tolerating the wide variety of salinities found in estuarine waters (Gerry 1981). Larval spot in Tampa Bay were collected in waters with salinities ranging from 6-32 ppt (Peters and McMichael, umpublished

Table 3-14. Acute toxicity of selected toxicants to various life stages of spot. Life stage: E = eggs; L = larvae; J = juveniles; A = adults; NR = not reported. Where salinity is not given, tests were conducted in saline water.

Substance	Life Stage	Salinity (ppt)	Temperature (°C)	Effect	Concentration (μgL^{-1})	Reference
Inorganic						
Cadmium	L	16-19	15-22	96-h LC_{50}	31	Middaugh et al. 1975
Copper	E		17	50% Mort. (24 h)	101*	Engle and Sunda 1979
Organic						
DDE	J	26	12	48-h LC_{50}	>100	Foster 1987
Dieldrin	J	25	12	24-h LC_{50}	3.2	Parrish et al. 1973
Mirex	J	27	22	48-h LC_{50}	>2000	Foster 1987

* Measured as cupric ion activity

Source: Homer and Mihursky 1991

manuscript 1992). Juveniles have been collected in Tampa Bay in salinities of 0-35 ppt (FIMP unpublished data, 1992).

Spot enter lower salinity backwaters and tidal tributaries at post-larval and juvenile stages. Peebles et al. (1992) reported that the first life history stage of spot which had higher abundance in the Little Manatee river versus Tampa Bay occurred during the post larval stage. Juvenile spot were most abundant in the oligohaline to polyhaline portions in the Little Manatee River, and much less abundant in the freshwater regions (Haddad et al. 1992). Spot were less abundant in McKay Bay (in Tampa Bay) during periods of low salinity. Naughton and Saloman (1978) also found spot to be more abundant in high salinity areas as compared to low salinity areas or St. Andrew Bay, Florida. Similar distribution patterns occur in the Chesapeake Bay area. Spot occur primarily in brackish to high salinity areas, and less frequently in freshwater regions (Homer and Mihursky 1991).

For many estuarine species, occupation of low salinity habitats is followed by the gravitation of larger individuals toward higher salinities (Peebles et al. 1992). In the Little Manatee River, these authors noted a positive correlation between spot mean length and higher salinities, suggesting a return to higher salinity waters with growth. Miller et al. (1984) suggested that spot migrate from their primary nursery grounds due to a decreased low salinity tolerance as the fish age. However, some studies have reported that size distribution of spot is independent of salinity (Reid and Hoese 1958; Parker 1971), suggesting that further research is necessary to fully document salinity requirements of spot life stages.

Hodson et al. (1981b) reported that both salinity requirements and water temperature change may prompt migration of larger juvenile spot into Gulf of Mexico waters. These authors also noted that effects of salinity and temperature interactions are often complex and difficult to interpret for this species.

3.8.4.2 Temperature

Spot are a eurythermal species and have been reported in water temperatures ranging from 1.2-35.5°C (Hedgepath 1967; Parker 1971; Moser and Gerry 1989). In laboratory studies, spot eggs hatched within 48 hours and yolk sac was absorbed within 5 days at 20°C (Powell and Gordy 1980). Hettler et al. (1978) reported that upper and lower lethal hatching temperatures of spot were 28 and 14°C, respectively. Parker (1971) reported that juvenile spot are well adapted to temperatures in the 6-20°C range, but fish approaching one year or older were noticeably absent from collections at temperatures below 10°C. According to Dawson (1958), the lower lethal water temperature of spot is between 4 and 5°C, and is dependent upon the size of the fish, rapidity of temperature drop and duration of exposure. Hoss et al. (1988) reported that larval spot show signs of thermal stress at water temperatures at and below 10°C. Continued exposures to low temperatures eventually resulted in mortality. This study suggests that low water temperatures during the larval stage could be detrimental to spot survival during recruitment into estuaries. Extreme water temperature drops associated with sudden freezing temperatures have led to mass mortality of some species during early recruitment (Lewis and Estevez 1988). Joseph (1972) noted large annual fluctuations in spot year class strength and reported that these non-periodic fluctuations are most likely due to environmental conditions on the spawning grounds. These studies suggest that early life stages of spot could be susceptible to low water temperatures (9-10°C) and that fluctuations in water temperatures could affect recruitment during some years.

No juvenile or adult spot were reported in cold water fish kills in the Tampa Bay estuary during 1962 or 1977 where water temperatures as low as 9.8°C were reported (Rinckey and Salomen 1964; Gilmore et al. 1978). Several studies suggest that seasonal water temperature declines trigger a migration into deeper portions of estuaries or into offshore waters (Pacheco 1962; Hodson et al. 1981b; Chittenden 1989; Rothschild 1990).

Several studies have reported upper thermal tolerance levels for spot. In North Carolina estuaries, juvenile spot had critical thermal maxima ranging from 31-38°C. (Bridges 1971; Horton and Bridges 1973; Hoss et al. 1972; Hoss et al. 1973). Spot taken from Galveston Bay, TX had 3-hr LD_{50}s at 35 and 36°C and 30-min LD_{100}s at 36-38°C (Chung 1977). All size classes of spot appeared to prefer water in the 25-34°C range and avoided water of higher temperatures (Galloway and Strawn 1974). In laboratory experiments, post larval and juvenile spot ($<$ 25 mm SL) had an upper lethal temperatures of 35.2°C, at salinities of 20 ppt, although they could tolerate higher temperatures at lower salinities (Hodson et al. 1981b). These studies suggest that average water temperatures in the Tampa Bay estuary, which generally range from 11-32°C (Lewis and Estevez 1988), are usually suitable for spot.

3.8.4.3 Dissolved Oxygen

Available information suggests that spot are very tolerant of low dissolved oxygen conditions. In the Chesapeake Bay they have been reported from areas with DOs less than 2 mg/L (Thorton 1975; Burton et al. 1980; Rothschild 1990) but are most common where DO's exceed 4 mg/L (Markle 1976; Chao and Musick 1977; Rothschild 1990; cited from Homer and Mihursky 1991). Many prey items of spot are not tolerant of low dissolved oxygen conditions (< 2 mg/L) and therefore low dissolved oxygen may indirectly effect their distribution patterns (Vernberg 1972). A more detailed analysis of the FDNR's FIMP database could potentially characterize preferred oxygen requirements of spot in Tampa Bay.

Burton et al. (1980) reported that juvenile spot (at 28°C) had 1 and 96 hour LC_{50}s of 0.49 and 0.70 mg/L, respectively. One and 96-hour LC_{95}s were 0.43 and 0.60 mg/L, respectively. Oxygen consumption by spot increases with fish weight, swimming speed and activity. They appear to be more efficient oxygen consumers than striped bass or white perch (Neumann et al. 1981; cited from Phillips et al. 1989).

Bay, there is a perceived movement of young spot from inshore to deeper estuarine and offshore waters with growth (Peters and McMichael, unpublished manuscript 1992).

3.8.4.5 Structural Habitat

Peters and McMichael (unpublished manuscript 1992) reported structural habitat preferences for postlarval and juvenile spot. Pelagic juvenile spot (> 8 mm) enter the Tampa Bay estuary and school over seagrass beds and near structures such as rocks or seawalls. This structured habitat appeared important for postlarval fish during the transition between a planktonic and benthic stages. Spot less than 20 mm were occasionally collected on sandy beach areas inside passes. They were common in seagrass beds, and abundant along seawalls. Fish > 20 mm were abundant throughout the bay in both seagrass and backwater areas, but were rarer around seawall areas. Juveniles 20-70 mm were abundant throughout the Tampa Bay estuary. Abundance of juvenile spot collected in backwater areas and seagrass beds were similar, suggesting that these fish had little preference between habitat types (Peters and McMichael, unpublished manuscript 1992). Homer and Mihursky (1991) noted that spot show little habitat preference with the exception of substrate preferences. Spot appear to prefer mud or mud sand mixtures of sediment. Prey abundance within these sediments apparently controls the distribution of spot with respect to substrate. They are capable of feeding much more efficiently in soft substrates (Smith and Coull 1987).

Environmental requirements of spot are summarized in Table 3-15.

3.9 STRIPED (BLACK) MULLET *(Mugil cephalus)*

The striped mullet is distributed throughout the world oceans in tropical, sub-tropical and temperate waters. It is generally found between 42°N and 42°S latitudes (Thomson 1963). In the western Atlantic it ranges from Brazil to Nova Scotia, (including the Gulf of Mexico) and is very common along Florida's west coast (Hoese and Moore 1977).

Striped mullet inhabit estuarine environments as well as freshwater and coastal marine areas (Broadhead 1953; Hendricks 1961; Thomson 1963, 1966). With the exception of annual spawning migrations, they generally remain in estuaries throughout the year in the west central Atlantic and Gulf of Mexico (Broadhead 1953).

Striped mullet is the most important inshore commercial finfish in Florida, contributing approximately 25% to the total finfish landings. Eighty-two percent of the total mullet production comes from the west central Florida region (Mahmoudi, pers. comm. 1992). The mullet fishery recently increased in importance due to the development of a multi-million dollar export market for roe (Mahmoudi, pers. comm. 1992). Commercial striped mullet harvest in Tampa Bay was on the order of 5.5-6.5 million pounds per year from 1986 through 1990 (Brown, pers. comm. 1992).

Table 3-15.

A =adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (^oC)				1.2-35.5	Hedgepath 1967; Parker 1971; Moser and Gerry 1989
	25-34^{o}C				Galloway and Strawn 1974
		14 (E)	28(E)		Hettler and Clements 1978
				6-20 (J)	Parker 1971
		10 (A,L)			Parker 1971;Hoss et al. 1981
		4-5			Dawson 1958
			31-38 (J)		Bridges 1971; Horton and Bridges 1973; Hoss et al. 1972, 1973
			35-36 3 hr LD_{50} (J)		Chung 1977
			36-38 30 min LD_{50} (J)		Chung 1977
Salinity (ppt)				0-60	Hedgepath 1967; Johnson 1978
				6-32 (L)	Peters and McMichael, unpublished ms.
				0-35 (J)	Peters and McMichael, unpublished ms.
	oligohaline to marine				Haddad et al. 1992; Homer and Mihursky 1991

	Preferred	Lower Umit	Upper Limit	Range	Reference
Dissolved Oxygen (mg/L)	**>4 (J,A)**			to <2 (J,A)	Thorton 1975; Burton et al. 1980; Rothschild 1990
	>4 (J,A)				Markle 1976; Chao and Musick 1977; Rothschild 1990
			0.49 (J) **1 hr LC_{50}**		Burton et al. 1980
			0.70 (J) **96 hr LC_{50}**		Burton et al. 1980
Depth (m)	<2 (J)				Springer and Woodburn 1960; Peters and McMichael,
	>2 (J,A)				Coastal Environmental Services 1992
				seasonaland dependent on **life stage**	Peters and McMichael unpublished ms.
Substrate	structure rocks, seagrass (L,J)				Peters and McMichael, unpublished ms.
	muddy sediments (J,Al)				Homer and Mihursky 1991
				mud-sand(J,A)	FlMP 1989, 1990

Striped mullet is the most important inshore commercial finfish in Florida, contributing approximately 25% to the total finfish landings. Eighty-two percent of the total mullet production comes from the west central Florida region (Mahmoudi, pers. comm. 1992). The mullet fishery recently increased in importance due to the development of a multi-million dollar export market for roe (Mahmoudi, pers. comm. 1992). Commercial striped mullet harvest in Tampa Bay was on the order of 5.5-6.5 million pounds per year from 1986 through 1990 (Brown, pers. comm. 1992).

3.9.1 LIFE HISTORY

Striped mullet spawning in Florida occurs primarily from October through February, peaking in December (Broadhead 1953). Mullet spawning has been reported to occur nearshore (Breder 1940), along beaches (Gunter 1945) and offshore (Broadhead 1953). However, offshore spawning appears to be dominant along the west coast of Florida (Broadhead 1953; Arnold and Thompson 1958; Mahmoudi, pers. comm. 1992), with spawning observed as far as 65-80 km offshore (Arnold and Thompson 1958). Egg and early larval striped mullet have been collected as far as 280 km offshore of the Tampa Bay estuary (Mahmoudi, pers. comm. 1992), suggesting that spawning had recently occurred at that location.

Planktonic larvae migrate toward inshore waters and estuaries where early juveniles use shallow water habitats as nursery areas. They arrive in estuaries at 4-6 weeks of age and approximately 20-25 mm SL (Major 1978; cited from Edwards 1990). Preliminary information suggests that striped mullet take approximately 1.5 months to reach Tampa Bay from offshore spawning grounds (Mahmoudi, pers. comm. 1992). Striped mullet larvae and early juveniles recruit into Tampa Bay from December through May. Recruitment peaked in late February and early March during 1987 and 1988 in Tampa Bay (Mahmoudi, pers. comm. 1992).

Juvenile striped mullet are abundant in tidal tributaries during winter months. Edwards (1990) reported that post-larval striped mullet (< 20 mm) recruited to the Manatee River during early January. From January to February, these juvenile striped mullet increased in size to 20-40 mm SL. Recruitment also peaked during the winter months in the Little Manatee River system (Haddad et al. 1992). In the Alafia River, juvenile striped mullet were abundant from January through early spring months, after which time they were collected much less frequently (Table 3-16; FIMP 1989, 1990). It is not known whether this is a result of migration from the backwater habitats where sampling was conducted or of increased gear avoidance due to increasing size. Springer and Woodburn (1960) reported that decreased catches of striped mullet > 80 mm may be due to gear avoidance.

Adult mullet occupy all regions of the Tampa Bay estuary including tidal tributaries; however, they prefer shallow waters (FIMP 1989; Coastal Environmental Services 1992). Tagging studies have documented that adult striped mullet tend to migrate very little and show little inter-estuary movement. Broadhead and Mefford (1956) reported that along the west coast of Florida, 70.3% of striped mullet stayed within 5 miles of the tag release location, 90% stayed within 20 miles, and 95% within 50 miles of release site. They

Table 3-16. Abundance and average size of juvenile striped mullet collected at two Alafia River stations during 1990. Source: FIMP 1990.

Station 1 Canal			Station 2 Alafia River Cove		
Month	Mean Density (# fish/100 m^2)	Avg Size (mm)	Month	Mean Density (# fish/100 m^2)	Avg Size (mm)
Jan	6.68	33	Jan	11.63	21
Feb	9.47	41	Feb	40.85	34
Mar	12.59	39	Mar	23.87	35
Apr	1.29	46	Apr	11.34	38
May	3.68	60	May	1.41	▪
			Jun	3.71	50

- May collections contained subadult and adult fish, therefore, average size not representative of juveniles.

tended to remain within the same bay system in which they were released. In the Tampa Bay system, adult mullet generally moved toward the mouth of the bay during September through December, then either directly offshore or north along the coastal shore until leaving for offshore waters to spawn. Mullet return to their original bay system after spawning and move into shallow, protected waters during spring months, where they remain until the following spawning season (Mahmoudi, pers. comm. 1992).

Age and growth rates of striped mullet have been examined in the Tampa Bay estuary. Juveniles grow an average of 100 mm their first year, while adult growth is generally between 40-60 mm per year, decreasing with increasing age of the fish (Mahmoudi, pers. comm. 1992). Maximum lengths, as estimated from the von Bertalanffy growth equation, are 497 and 372 mm FL for females and males, respectively (Mahmoudi, per. comm. 1992). Striped mullet mature at 1 to 3 years of age, and males tend to mature at smaller sizes (younger ages) than females (Broadhead 1953; Mahmoudi, pers. comm. 1992). Age structure analyses suggested that three-year olds were the dominant group in Tampa Bay. Average length at age for Tampa Bay striped mullet (sexes combined) was 186, 267, 313, 350, and 387 mm FL for age 0+ through age 4+, respectively (Mahmoudi, pers. comm. 1992).

3.9.2 ECOLOGICAL ROLE

Striped mullet are one of the few large marine fishes that feed directly on the first trophic level, living and dead plant matter (Collins 1985). They are an ecologically important component in the flow of energy through estuarine communities (Collins 1985). Diet and feeding behavior of mullet may vary by location but their food is primarily comprised of epiphytic and benthic microalgae, macrophyte detritus, or inorganic sediment particles. Sediment particles function as a grinding paste in the gizzard-like pyloric portion of the stomach, although some small particles are rich in adsorbed micro-organisms and are selectively ingested for their food value (Odum 1970). Larval and post larval mullet feed on zooplankton (Nash et al. 1974). In Indian River, Florida, larvae up to 15 mm fed entirely on copepods (70%) and mosquito larvae (30%), larvae 15-25 mm SL fed on copepods (50%), mosquito larvae (15%) and plant detritus (35%), and juveniles 25-35 mm fed primarily on plant detritus (80%) and copepods (10%) (Harrington and Harrington 1961). Mahmoudi (pers. comm. 1992) reported that at approximately 50 mm, juveniles become strictly herbivores. Adult striped mullet are herbivorous or iliophagous, feeding either by taking detrital and other organic matter from the sediment or by grazing off epiphytic and filamentous algae (Thompson 1966).

3.9.3 ENVIRONMENTAL REQUIREMENTS

Numerous references are available on the salinity and temperature requirements of striped mullet, however, very few are specific to Tampa Bay. Other environmental requirements such as dissolved oxygen, water depth and structural habitat may also play a role in striped mullet distribution and abundance throughout the Tampa Bay estuary, and these will also be discussed in the following sections.

3.9.3.1 Salinity

In laboratory tests, striped mullet egg survival was highest at near seawater salinities (Sylvester et al. 1975; Fig. 3-29). In tests comparing salinities of 24-32 ppt, highest egg survival rates were at 32 ppt. (highest salinity tested) whereas greatest larval survival was at 26 ppt. This supports the assumption that spawning occurs in marine waters and that the earliest life stages are stenohaline marine forms. Lee and Menu (1981) tested effects of salinity on mullet egg development and hatching in Hawaii. These authors found that striped mullet eggs developed to embryonic stage at salinities of 5-60 ppt and hatching occurred at all salinities between 10 and 55 ppt. Optimum salinity for eggs (at 22-25.5°C) was 30-40 ppt, with a peak at 35 ppt.

Nordlie et al. (1982) examined the osmoregulatory capabilities of three size classes (20-29, 30-39 and 40-69 mm SL) of juvenile striped mullet in six salinities (freshwater to saltwater). These authors reported that the two smaller groups could tolerate instantaneous transfer from brackish (6-23 ppt) to all salinities but fresh, while the 40-69 mm size group tolerated all salinities. From these results, these authors suggested that osmotic regulatory capacities become independent of body size as the fish grows. Adult mullet are euryhaline and have been reported in waters with salinities ranging from 0 (Collins 1981) to 75 ppt (Simmons 1957).

Juvenile striped mullet are found throughout Tampa Bay in waters with salinities ranging from 0.3 to 35 ppt (Springer and Woodburn 1960; FIMP 1989, 1990; FIMP, unpublished data). They appear to prefer low salinity riverine systems, and were reported in high abundance in the Alafia, Little Manatee, and Manatee river systems (FIMP 1989, 1990; Edwards 1990; Haddad et al. 1992). However, large catches in McKay Bay (Price and Schlueter 1985), an area characterized by more polyhaline salinities, suggest that salinity may not be the sole factor influencing their distribution within the estuary.

In the Manatee River, striped mullet early juveniles (20-40 mm) were significantly more numerous (83% of 716 fish) at stations where salinities ranged from 16-20 ppt and none were caught in salinities less than 5 ppt. The fact that these small juveniles were more abundant at stations with higher salinities supported studies which suggested that complete osmoregulatory capacity and ability to tolerate freshwater conditions may not be attained until around 40 mm SL (Nordie et al. 1982). However, ichthyofaunal collections in the Little Manatee River showed that peak immigration of striped mullet into the river occurred during the early juvenile stage (Peebles et al. 1992). Extensive juvenile finfish surveys conducted throughout the Little Manatee River system (polyhaline to freshwater) indicated that juvenile striped mullet were generally more abundant between river kilometer (rkm) 5.8 and 11.8 (Fig. 3-30). Salinity zones at these stations of maximum abundance ranged from mesohaline (5-18 ppt) to oligohaline (0.5-5) (Fig. 3-31; Haddad et al. 1992).

It appears that the riverine areas of Tampa Bay are important nursery areas for juvenile mullet, and that their distribution may be contingent upon salinity patterns within these areas. However, there is some inconsistency in literature relating to the preferred salinities of juveniles in their nursery habitats. It is possible that factors other than salinity contribute to striped mullet habitat selection within the Tampa Bay estuary.

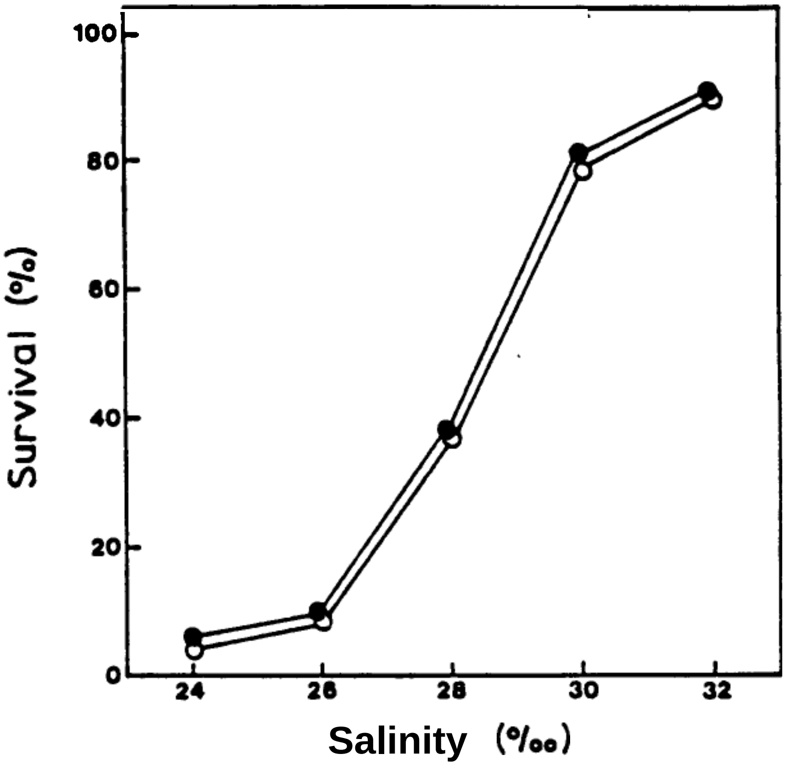

Figure 3-29. Survival of striped mullet eggs at different salinities for exposure periods of 24 h (●–●) and 48 h (○–○). Source: Sylvester et al. (1975).

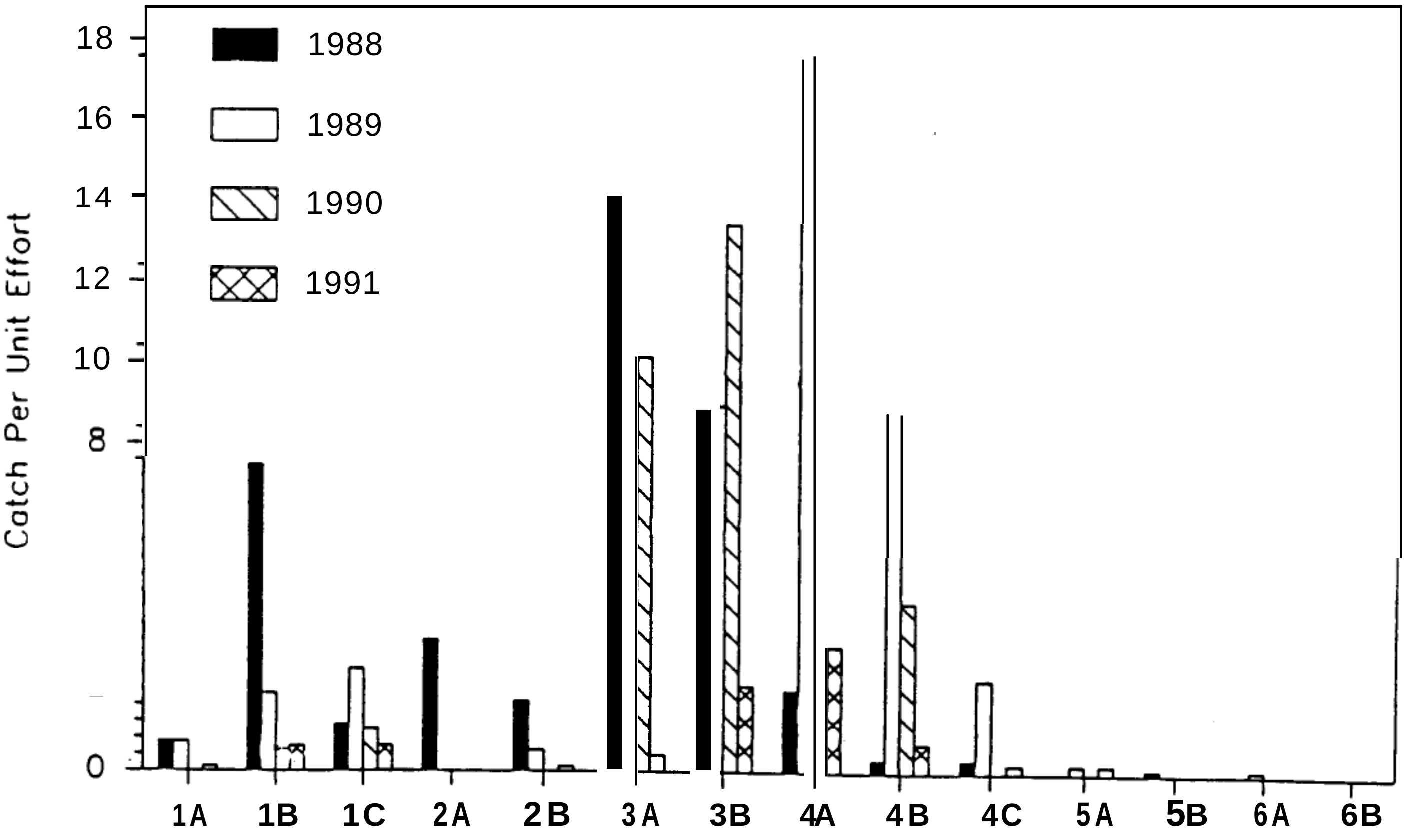

Figure 3-30. CPUE (# fish/100m^2) for juvenile striped mullet by sampling location in the Little Manatee River. Station 1 = rkm 0; station 2 = rkm 2.5; station 3 = rkm 5.8; station 4 = rkm 11.8; station 5 = rkm 15.5; station 6 = rkm 16.4. Source: Haddad al. 1992.

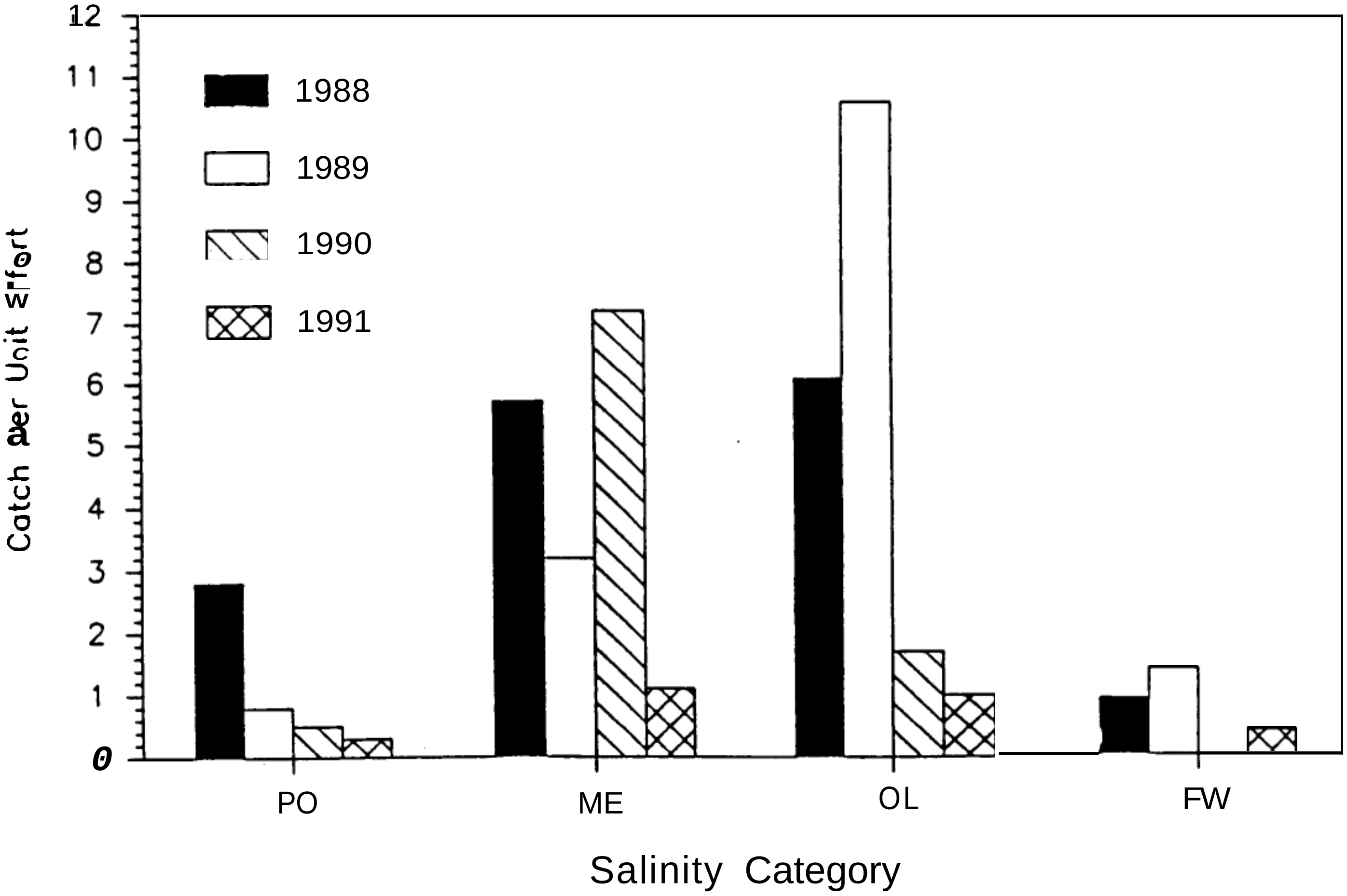

Figure 3-31. CPUE (# fish/100m^2) for juvenile striped mullet by surface salinity in the Little Manatee River. PO = polyhaline (> 18 ppt); ME = mesohaline (5-18 ppt); OL = oligohaline (0.5-5 ppt); and FW = freshwater (0-0.5 ppt). Source: Haddad et al. 1992.

3.9.3.2 Temperature

Nash and Sylvester (1974) investigated thermal tolerances of egg and larval striped mullet in Hawaii. They found that mullet survived over broad temperature ranges (10.3 to 31.9°C). Minimal mortalities for both eggs and larvae occurred between 18.9 and 25.3°C. Along Florida's Gulf coast, juvenile striped mullet were found at temperatures from 13°C to 34.5°C (Kilby 1949). Springer and Woodburn (1960) collected striped mullet in Tampa Bay waters at temperatures of 10.7 to 32.5°C. They reported that these temperatures did not appear to limit the distribution of adult or juvenile striped mullet.

Upper thermal tolerance limits of juvenile striped mullet have been investigated in Galveston Bay, Texas. Fish 80-280 mm long had 3 hr LD_{50}s at 37-38°C and 30 min LD_{100}s at 39-41°C (Chung 1977). The critical thermal maximum (CTM) of Hawaiian mullet (70-125 mm SL) was affected by acclimation temperature and time of day (Sylvester 1975). Mean CTM increased from 38.5 to 41.3°C for fish acclimated at 20° and 29°C, respectively.

Striped mullet are apparently susceptible to severe cold water conditions. They have been reported in cold water fish kills in Tampa Bay (Gilmore et al. 1978) where water temperatures dropped below 10°C in many areas. Juveniles (10-25 mm SL) may be less sensitive to cold than adults. Juveniles in the Indian River area were captured in active schools where water temperature was 10°C; however, adult mortality was observed at the same time (Gilmore et al. 1978). Juveniles were also very abundant during winter months in shallow tidal tributaries where low water temperatures were prevalent (FIMP 1990; Edwards 1990; Haddad et al. 1992; Fig. 3-32).

3.9.3.3 Dissolved Oxygen

Mullet eggs and larvae are very sensitive to low dissolved oxygen (D.0.) conditions. Sylvester et al. (1975) stated that mullet larvae apparently cannot survive where D.O. falls below 4 ppm. Striped mullet eggs incubated in the lab for 48 hrs (32 ppt. salinity) had a survival rate of 0 to 3% at D.O. concentrations of 4.5 ppm and below, and 85-90% at D.O. concentrations of 5 ppm and above (Sylvester et al. 1975). Over a D.O. range of one to eight ppm, median egg survival was between 4.5 and 5 ppm for 48 hrs and median larval survival occurred between 6.4 to 7.9 ppm for 96 hrs (Fig. 3-33). Walsh et al. (1989) found that oxygen consumption rates of laboratory reared, larval striped mullet did not vary significantly among salinities ranging from 10-35 ppt, but increased temperatures elicited significant increases in oxygen consumption. No specific dissolved oxygen requirement information was available for juvenile or adult striped mullet.

3.9.3.4 Water Depth

Striped mullet live in a wide range of water depths and spawn primarily in relatively deep cool coastal waters along the west coast of Florida (Arnold and Thompson 1958). Early larvae (< 3 mm) were primarily collected in waters of 100-1000 fathoms at a distance of 160-280 kilometers offshore the Tampa Bay estuary (Mahmoudi, pers. comm.

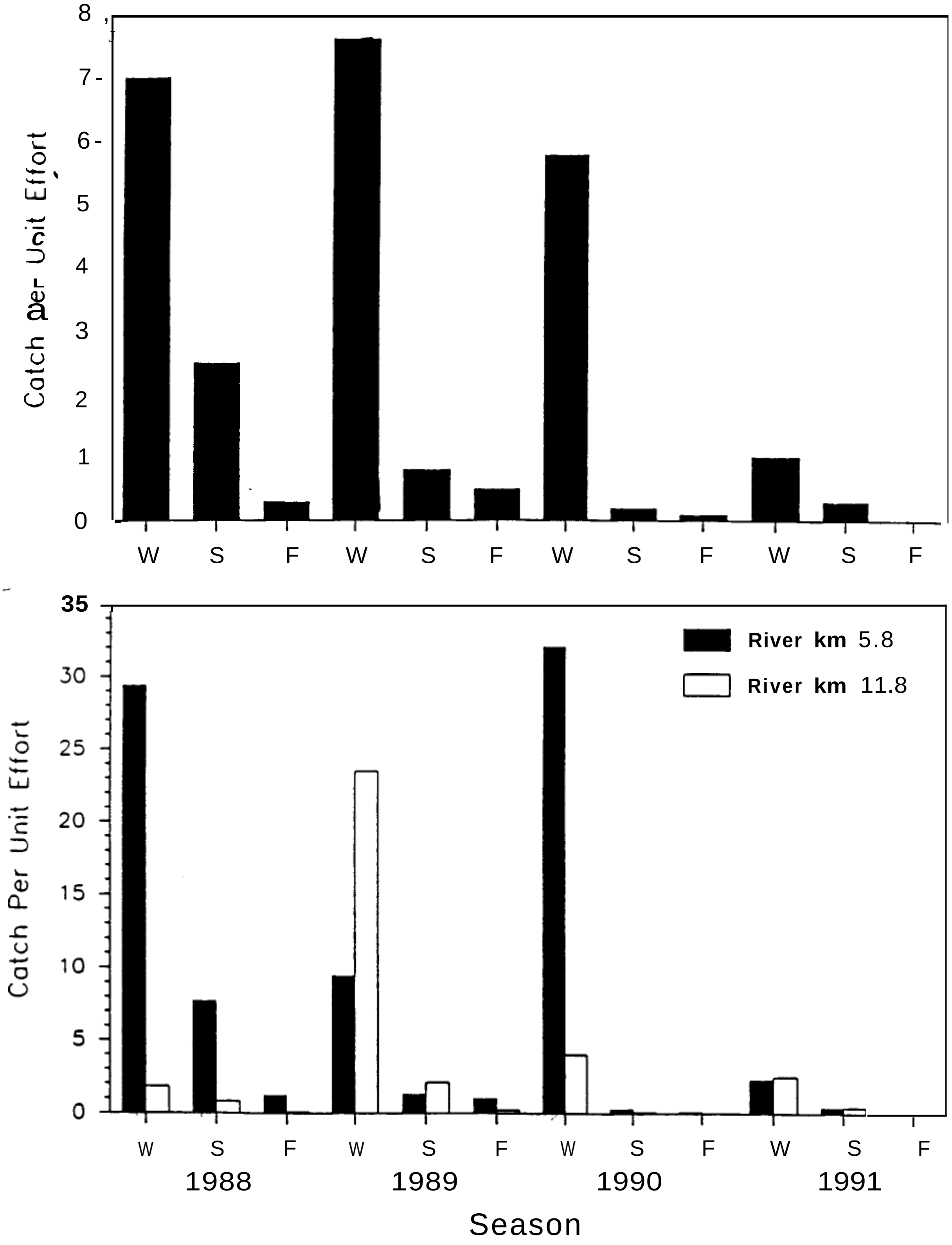

Figure 3-32. CPUE (# fish/100m^2) for juvenile striped mullet by season in the Little Manatee River during 1988-1991. W = winter (Jan-Apr); S = summer (May-Aug); F = fall (Sep-Dec). Source: Haddad et al. 1992.

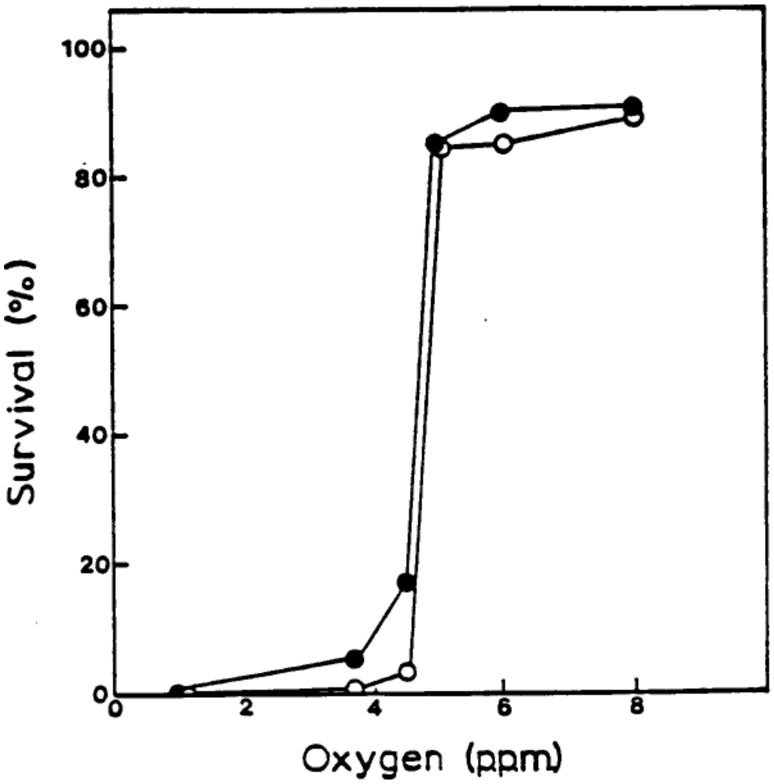

Figure 3-33. Survival of striped mullet eggs at different oxygen concentrations for exposure periods of 24 h (●–●) and 48 h (○–○) at 32% salinity. Source: Sylvester et al. 1975.

1992). The larvae then move inshore to estuarine waters (Collins 1985). Striped mullet larvae entering Tampa Bay exhibited a significant depth effect with respect to their mean size. Larvae were significantly larger in surface waters than in mid or bottom waters (Robison 1985). Juveniles in Tampa Bay prefer shallow habitats, as evidenced by their frequent capture with seines, which generally sample water less than 2 meters in depth (FIMP 1989, 1990). Edwards (1990) reported that striped mullet use shallow habitats like tidal creeks and creek mouths (< 2 m) within the Manatee River System. Adults were caught in shallow waters (< 2 m) throughout the Tampa Bay system (FIMP 1989; Coastal Environmental Services 1992).

3.9.3.5 Structural Habitat

Egg and larval stages of striped mullet are pelagic and are probably more dependent on specific water quality requirements rather than structural habitat. Juvenile mullet utilize a variety of habitats within the Tampa Bay estuary, however they appear to prefer tidal tributary systems. Densities reported from these areas are higher than those reported in many main bay locations (FIMP 1989, 1990). Early juvenile mullet prefer habitats such as tidal creeks and creek mouths in the Manatee River. They were rarely collected from linear mangrove habitats and marsh shorelines (Edwards 1990). In the Little Manatee River and Alafia Rivers, large collections of striped mullet occurred along both linear marsh shorelines and in more specialized habitats like coves, canals and other backwater areas (FIMP 1989, 1990; Haddad et al. 1992). Shorelines at these stations ranged from steep banks with overhanging trees and terrestrial vegetation to more gently sloping shorelines with emergent vegetation such as *Rhizophora mangle, Spartina sp.,* and *Juncus sp.*

Large numbers of juvenile mullet were typically collected over muddy or mud/sand bottom substrates with no attached bottom vegetation. Juvenile striped mullet were very abundant in McKay Bay, an area characterized by bare sand and silty sediments. (Price and Schlueter 1985). Similarly, unvegetated, muddy, or mud/sand bottoms are present in the tidal tributaries, where juveniles are common.

Very little data are available to assess critical habitats of adult mullet. Except for spawning runs, they apparently remain year round in estuaries. Adults were collected throughout Tampa Bay in gillnet samples over bare and vegetated bottoms during spring and fall 1989 (FIMP 1989).

Environmental requirements of striped mullet are summarized in Table 3-17.

3.10 CLOWN GOBY *(Microgobius gulosus)*

3.10.1 INTRODUCTION

Gobiid fishes are common in shallow, estuarine waters of subtemperate and tropical regions (Darcy 1980). The clown goby ranges from the Chesapeake Bay south along the

Table 3-17. General and preferred ranges and upper and lower tolerance limits for environmental requirements of striped mullet. Letters in parentheses indicate life stage. S = spawning, E = egg, L = larval, J = juvenile, A = adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)	18.9-25.3 (E,L)			10.3-3.9 (E,L)	Nash and Sylvester 1974
				13-34.5 (A,J)	Kilby 1949
				10.7-32.5 (A,J)	Springer and Woodburn 1960
			37-38 3 hr-LD_{50} (J)		Chung 1977
			39-41 30 min-LD_{100} (J)		Chung 1977
		10 (A)			Gilmore et al. 1978
Salinity (ppt)	>32 (E)				Sylvester et al. 1975
	26 (L)				Sylvester et al. 1975
	30-40 (E)			10-55 (E)	Lee and Menu 1981
	0.5-18 (J)				Haddad et al. 1992
				0-75	Collins 1981; Simmons 1957
				0.3-35 (J)	FIMP unpubl. data
Dissolved Oxygen (mg/l)		4 (L)			Sylvester et al. 1975
		54.5 (E)			Sylvester et al. 1975
Depth (m)	surface waters (E,L)				Arnold and Thompson 1988
	< 2 m (J)				FIMP 1989, 1990
Substrate	mud, mud-sandy (J)				FIMP 1989, 1990; Haddad et al. 1992

Atlantic coast and along the Gulf of Mexico to Corpus Christi, Texas (Hoese and Moore 1977).

The clown goby is not considered an economically important species, however, it functions as an important ecological link within the Tampa Bay ecosystem. This species has been identified as prey for juvenile spotted seatrout (McMichael and Peters 1989) and is probably prey for numerous other carnivorous fish species.

3.10.2 LIFE HISTORY

Little information is available to describe the life history of the clown goby in the Tampa Bay estuary. Larval and postlarval *Microgobius* spp. (primarily *M gulosus)* are present year-round in Tampa Bay near the Little Manatee River. Peak spawning occurs during spring and summer months (Peebles, in prep.) Clown goby life history information was also available for the Indian River area of Florida (Provancha and Hall 1991a). These authors reported that clown goby spawning was initiated between the months of March and May. Early juveniles (< 15 mm SL) were collected during May. This species appeared to have a protracted spawning period which continued through late fall. Reid (1954) suggested a protracted spawning season for *M gulosus* on the west coast of Florida, near Cedar Key. Kilby (1955) and Birdsong (1981) suggested that spawning may occur year round in south Florida estuaries. Springer and Woodburn (1960) reported ripe individuals in Tampa Bay as late as November, suggesting that spawning could potentially occur year round in this estuary. Provancha and Hall (1991a) suggested that clown goby fecundity was low, with a mean number of 305+-77.5 ova for females 35-49 mm TL.

Juvenile and adult clown goby occur in shallow (<2 m) regions of the Tampa Bay estuary (Haddad et al. 1992). They appear to prefer vegetated seagrass areas to non-vegetated bottoms. More information is necessary to fully and accurately describe life history parameters of this species. No information was available on size or age at maturity or longevity. Juvenile clown goby grow to a size of approximately 35-40 mm during the first year and 50-60 mm TL during the second year in Indian River lagoon (Provancha and Hall 1991a). Growth after age two was less than 10 mm per year. Estimated mortality rates were 95% annually.

3.10.3 ECOLOGICAL ROLE

Studies on clown goby feeding habits suggest that they are primarily omnivorous. Odum (1971) provided quantitative stomach content analysis data for the clown goby in south Florida. He reported that fish 18-32 mm in length fed primarily on amphipods (43% by volume), harpacticoid copepods (21%) and chironomid larvae (10%). Food items of lesser significance included cumaceans, cladocerans, mysids, plant detritus and algal strands. Carr and Adams (1973) reported that juvenile clown goby on the west coast of Florida near Crystal River might be classified as detritivorous, since detritus, sand grains and fecal pellets accounted for 51 to 79% of injested material. However, they reported that the mixed nature of the diet was more indicative of an omnivore. Clown gobies 11-25 mm SL consumed primarily detritus and copepods. Fish 26-40 mm SL shifted to a diet of detritus and benthic invertebrates, especially amphipods and polychaetes (Fig. 3-34). Reid (1954) reported that larger clown gobies (45-57 mm) near Cedar Key, Florida fed primarily on mysids, copepods, and amphipods. Stomach contents of clown gobies (18.5-43.3 mm) from the Tampa Bay estuary contained copepods, gammarids, unidentified crustaceans, polychaetes, tiny bivalves and algae (Springer and Woodburn 1960).

3.10.3.1 Predators

Very little information is available to document predators of the clown goby in the Tampa Bay estuary. This species has been identified as a prey item for juvenile spotted seatrout (McMichael and Peters 1989; Peebles 1992). Considering the abundance of this species in the Tampa Bay estuary, it is likely that the clown goby may serve as potential prey for numerous species of predatory fishes.

3.10.4 ENVIRONMENTAL REQUIREMENTS

The clown goby is not considered to be an important commercial or recreational fishery species within the Tampa Bay estuary. As a consequence of this, very little research has been directed at identifying their specific environmental requirements. General information can be synthesized from fishery independent studies which have been conducted within the estuary.

3.10.4.1 Salinity

No data were available to document critical salinities of the clown goby. It appears that this species can tolerate freshwater conditions, however, no information was available to document tolerance to hypersaline conditions. Darcy (1980) summarized data on known salinity ranges of this species. Clown gobies were collected in Florida waters with salinities ranging from 0 to 36.6 ppt.

Springer and Woodburn (1960) noted that salinity did not appear to be a determining factor in clown goby distribution within Tampa Bay. Specimens were collected in a full range of salinities from freshwater to saltwater. They reported that moderately high salinities (20-30 ppt) appeared to be characteristic of areas of greatest abundance. Recent collections of clown gobies throughout Tampa Bay and into the freshwater tidal portions of the tributaries confirmed the euryhaline nature of this species (FIMP 1989,1990). Juvenile and adult clown gobies were commonly collected in all tidal portions of the Little Manatee River, from freshwater to saltwater (Haddad et al. 1990; Haddad et al. 1992). Juveniles and adults were collected in salinities ranging from 0.3 to 37 ppt in Tampa Bay (FIMP, unpublished data). Clown gobies were collected in salinities as high as 44 ppt. in shallow coastal regions at the mouth of the Tampa Bay estuary (Fonseca, unpublished data 1992).

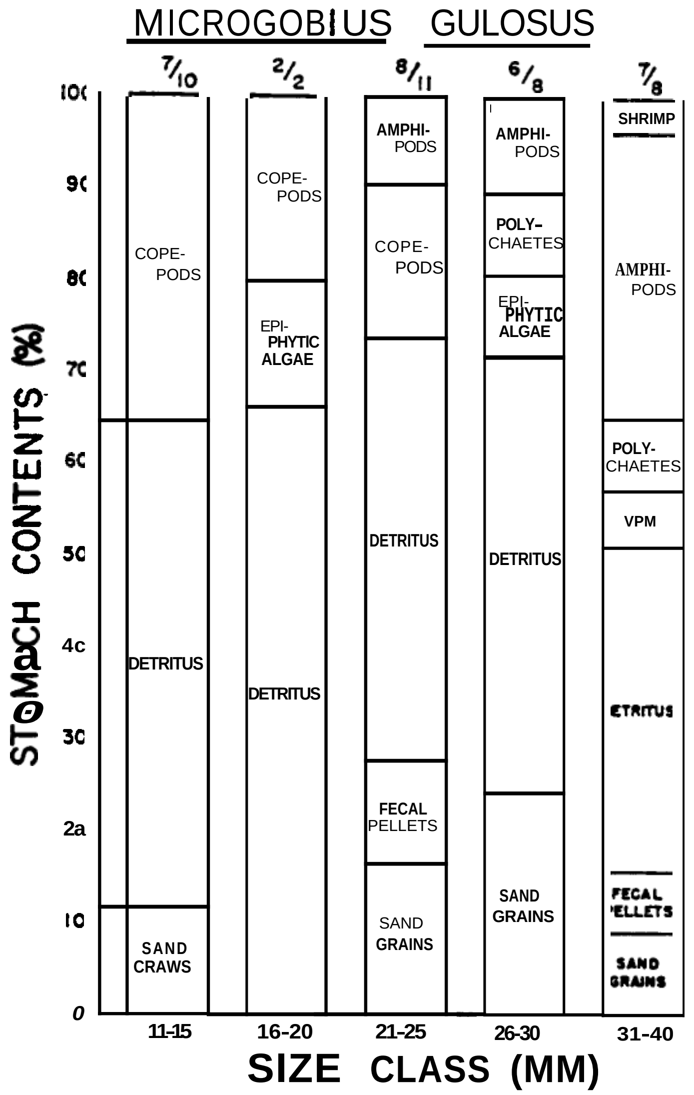

Figure 3-34. Stomach contents of juveniles of *Microgobius gulosus.* "VPM" = vascular plant material. Source: Carr and Adams (1973).

3.10.4.2 Temperature

Very little data are available to document temperature preferences or lethal limits of the clown goby. Darcy (1980) reported that clown gobies were collected in Florida waters with temperatures ranging from 12-34.1°C. Clown gobies were collected throughout Tampa Bay in water temperatures ranging from 14.8-31.6°C during Florida's Fisheries Independent Monitoring Program (FIMP, unpublished data). Cold water temperatures of approximately 10°C might impact the clown goby. A few "stunned" specimens (23-37 mm) were observed during cold water conditions in Tampa Bay during December, 1962 when water temperatures were reported to be 10.8oC (Rinckey and Saloman 1964); however, none were observed in a freeze which impacted the Tampa Bay area during 1977 when water temperatures were reported as low as 6-9oC (Gilmore et al. 1978). The cryptic benthic nature of this species may hinder mortality observations. No data are available to document their upper thermal tolerance levels.

3.10.4.3 pH

Space shuttle launches at Cape Canaveral, Florida have often caused acute acidification of adjacent estuarine waters which has lead to mortality in many fish species. Clown goby mortalities were commonly observed in shallow waters adjacent to the launch pad where pH values ranged from lows of 1.5 to normal values of 8.0-9.0 depending on distance from launch pad (Provancha and Hall 1991a). In Tampa bay, clown gobies were collected in waters with pH ranging from 4.3 to 8.5 (FIMP, unpublished data). Because of the buffering capacity of the Tampa Bay estuary, it is unlikely that they would be exposed to waters with pHs below these levels, except in the rare case of an acid spill.

3.10.4.4 Water Depth

Clown gobies primarily occur in the shallow water regions of estuaries. Kilby (1955) collected them in shallow pools (< 2 m) near Cedar Key, Florida. Reid (1954) collected clown gobies on shallow (< 1 m) sand flats and in deeper flats in channels. In Tampa Bay, the clown goby is typically collected in the shallow portions (< 2 m) of the estuary (Haddad et al. 1992).

3.10.4.5 Structural Habitat

The clown goby uses a variety of habitats within estuaries; however, they appear to prefer seagrass habitats versus bare bottoms and muddy bottoms versus coarser sediments. The clown goby was reported to be among the most numerically dominant species in seagrass beds in the Gulf of Mexico region (Gilmore 1987).

Juvenile and adult clown gobies are commonly collected in shallow seagrass habitats within Tampa Bay (Haddad et al. 1987, 1990, 1992; FIMP 1989, 1990). Springer and Woodburn (1960) reported that the clown goby appears to prefer muddy bottoms with vegetation, in protected water in Tampa Bay. Clark (1971) and Birdsong

(1981) suggested similar habitat preferences for this species in Whitewater Bay, Florida and Big Pine Key, Florida, respectively. Reid (1954) collected specimens from shallow sand flats, deeper flats, and channels. These shallow flats were vegetated with submerged aquatic vegetation including *Halodule wrightii, Syringodium filiforme, Thalassia testudinum* and *Ruppia maritima.*

Near Tarpon Key (lower Tampa Bay), clown goby densities in dense *Thalassia testudinum* ranged from 0.16 to 0.33 fish/m^2 (Haddad et al. 1987). Clown gobies were also commonly collected in shallow seagrass areas near the mouth of the Little Manatee River (Haddad et al. 1990). Collections containing the greatest numbers of these specimens were taken from moderate to dense stands of *Halodule wrightii,* however specimens were also collected in sparse *H. wrightii* and over bare sand (Haddad et al. 1990). Average densities for clown gobies collected in *H wrightii* beds during March through August 1990 were 0.33 fish/m^2 and this species comprised approximately 19% of the total drop net catch (Haddad et al. 1990). Clown gobies were commonly collected with drop nets in seagrass beds located throughout Tampa Bay during an extensive fisheries independent monitoring program (FIMP 1989, 1990). Average densities ranged to 0.87/m^2 and 3.06/m^2 during spring and fall 1989, respectively and to 0.60/m^2 and 1.13/m^2 during spring and fall 1990. The clown goby also has a ubiquitous distribution throughout the Little Manatee River system, from Tampa Bay upstream to the freshwater tidal region (Haddad et al. 1990). However, more information is necessary to assess specific structural habitat preferences within the river.

Preliminary information collected from a limited number of sample sites within Tampa Bay suggest that the clown goby may prefer areas of submerged aquatic vegetation characterized by higher shoot densities and more complex understory structures. They were more abundant in drop net collections in *Syringodium filiforme* and *Halodule wrightii* than in other vegetated habitats (e.g., *Thalassia, Caulerpa)* or bare areas. However, more information is necessary to accurately assess this hypothesis (Fonseca, unpublished data 1992).

These studies suggest that the clown goby has the ability to use a variety of structural habitats and subhabitats within the Tampa Bay estuary. Provancha and Hall (1991) reported that given the wide diversity of structural habitats used by the clown goby, one criterion for habitat selection may include protection from disturbances such as constant wave action or strong currents.

Environmental requirements of the clown goby are summaried in Table 3-18.

3.11 LINED SOLE *(Achirus lineatus)* AND HOGCHOKER *(Trinectes maculatus)*

3.11.1 INTRODUCTION

The family Soleidae is represented by three species in the Tampa Bay area. Two of these species, the lined sole and hogchoker, are common in the Tampa Bay estuary.

Table 3-18. General and preferred ranges and upper and lower tolerance limits for environmental requirements of clown goby. Letters in parentheses indicate life stage. S= spawning E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				12-34.1 (J,A)	Darcy 1980
		10.8 "stunned"			
				14.8-31.6 (J,A)	FIMP unpubl. data
Salinity (ppt)				0-36.6 (J,A)	Darcy 1980
	20-30 (J,A)				Springer and Woodburn 1960
				to 44 (J,A)	**Fonseca unpubl. data, 1992**
				0.3-37 (J,A)	FIMP unpubl. data
Dissolved Oxygen (mg/l)					
Depth (m)	**<2 (J,A)**				Kilby 1955; Haddad et al. 1992
Substrate	seagrasses, muddy vegetated bottoms (J,A)				Reid 1954; Haddad et al. 1992; Springer and Woodburn 1960; FIMP 1989, 1990
				Bare areas to dense vegetation (J.A)	Fonseca unpubl. data, 1992

Both species are very similar in appearance; however, larval-lined sole can be distinguished from hogchoker by the presence of a perforated interbranchial septum. Also, juvenile and adult hogchoker lack a pectoral fin. The lined sole is widespread in distribution from Florida to Uruguay along the Atlantic coast, and along the Gulf of Mexico (Briggs 1958). This species has a ubiquitous distribution in estuarine areas (Reid 1954; Springer and Woodburn 1960; Tabb and Manning 1961). The hogchoker ranges from Massachusetts to Panama (Hoese and Moore 1977) and is common in estuaries and shallow Gulf waters. It is apparently more abundant in brackish waters than the lined sole. Neither of these species can be considered economically important to the Tampa Bay estuary, although some hogchokers are harvested and sold as "freshwater flounder" in aquarium stores (Hoese and Moore 1977).

3.11.2 LIFE.HISTORY

Peebles (in prep.) collected lined-sole larvae in Tampa Bay from April through November. Futch (1970b) described the early life history of the lined sole in the Tampa Bay estuary. Eggs and larvae (3 mm SL) were collected from May through November. Maximum abundance occurred in June and July, and corresponded with highest water temperatures. Spawning appeared to be confined to inshore coastal areas, less than six miles offshore, in depths of two to five fathoms (3.7-9.1 m). Mid-depth ichthyoplankton samples at the mouth of Tampa Bay contained many lined sole larvae during May and June, 1980. Post-larvae were found to be significantly more abundant in bottom waters than at other depths (Robison 1985). Lined sole larvae were the second most abundant larvae collected in ichthyoplankton samples north of the Port Manatee area during August 1975 (Conservation Consultants 1976) suggesting that these larvae are very common throughout the lower Tampa Bay estuary.

Larval and postlarval hogchoker are present in Tampa Bay from April through November (Peebles, in prep.). None were reported in ichthyoplankton samples at the mouth of the bay (Robison 1985). These studies suggest that hogchoker spawning probably occurs within the Tampa Bay estuary but outside of the tidal tributaries. Hogchoker enter tidal tributaries as postlarvae; move upstream and are primarily concentrated in the upper tidal river by early juvenile stage. Adults return to higher salinity waters for spawning. They may actually drift in the outgoing tides at night to aid in movement downstream (Peebles, in prep.).

Lined sole and hogchoker undergo a period of metamorphosis when they change from a dorso-ventrally oriented fish to a form more typically observed in flatfish. In Tampa Bay, this metamorphosis occurs at approximately 3.0 mm SL. At this size, the left eye begins to migrate toward the dorsal midline. Behavior of newly hatched, laboratory reared lined sole larvae was described by Houde et al. (1970). Larvae were almost neutrally buoyant and did not attempt to swim until two days after hatching. At six days (2.5 mm SL) lined sole larvae began to swim for short periods near the bottom with the body axis tipped to the left. At eight days (2.8 mm SL) some larvae rested on the tank bottom for several minutes. By the tenth day, larvae (3.15 mm SL) spent nearly one half the time on the tank bottom. By the twelfth day (3.6 mm SL) most larvae remained on the tank bottom (Houde et al. 1970). Houde et al. (1970) report of lined sole and hogchoker larvae are similar in most respects and both species are occasionally taken together in Tampa Bay collections. No data were available to describe age and growth of juvenile and adult lined sole or hogchokers.

Very little information to document life history parameters of juveniles and adults of these species. Lined sole are commonly collected in shallow seagrass habitats in the Tampa Bay estuary. Hogchoker are collected over both vegetated and non-vegetated bottoms, although appear to prefer non-vegetated habitats in oligohaline or fresh waters of tidal tributaries. Hogchokers may occur over 10 km upstream of the freshwater interface and are very resistant to displacement by freshwater discharge (Peebles, in prep.).

3.11.3 ECOLOGICAL ROLE

After hatching, larval lined sole can survive without food for approximately 3 - 3.5 days (Houde 1974). Stomach contents of Tampa Bay lined sole larvae contained gastropod protoconchs and small clams, and juveniles fed primarily on copepods (Futch 1970b). Adults fed primarily on gammarid amphipods and polychaetes (Springer and Woodburn 1960). Qualitative studies on the food habits of hogchoker indicate that it feeds primarily on benthic invertebrates, including polychaetes, small crustaceans, algal strands, chironomid larvae, and sand (Hildebrand and Schroeder 1928; Reid 1954; McLane 1955 as cited from Carr and Adams 1973). Hogchoker feeding habits have not been described in the Tampa Bay estuary; however, information was available from the Crystal River area of Florida (Carr and Adams 1973). Juvenile (18-35 mm) hogchoker fed mainly on polychaetes which accounted for 60-88% of the stomach contents throughout that size range. The dependence on polychaetes showed an apparent gradual increase with growth of this species (Fig. 3-35). Detritus consumption was maximal in the smallest size class and decreased in importance as a food item of larger juveniles (Carr and Adams 1973).

3.11.4 ENVIRONMENTAL REQUIREMENTS

Very little information exists to document the environmental requirements of lined sole and hogchoker in Tampa Bay. Most of the available information pertains to their distribution and preferred salinity and water temperatures. More data are necessary to thoroughly describe dissolved oxygen, water depth, and structural habitat requirements.

3.11.4.1 Salinity

Both the lined sole and hogchoker are euryhaline species although hogchoker prefer more fresh or oligohaline waters. Lined sole were collected in waters with salinities ranging from 4-35 ppt in Tampa Bay (Springer and Woodburn 1960). These authors collected 91 lined sole (12.6-68.5 mm) at stations throughout the estuary (except the Gulf beaches) in an extensive ichthyofaunal study in Tampa Bay. Hogchoker were collected in salinities ranging from 0-36 ppt in Tampa Bay (FIMP, unpublished data). Springer and

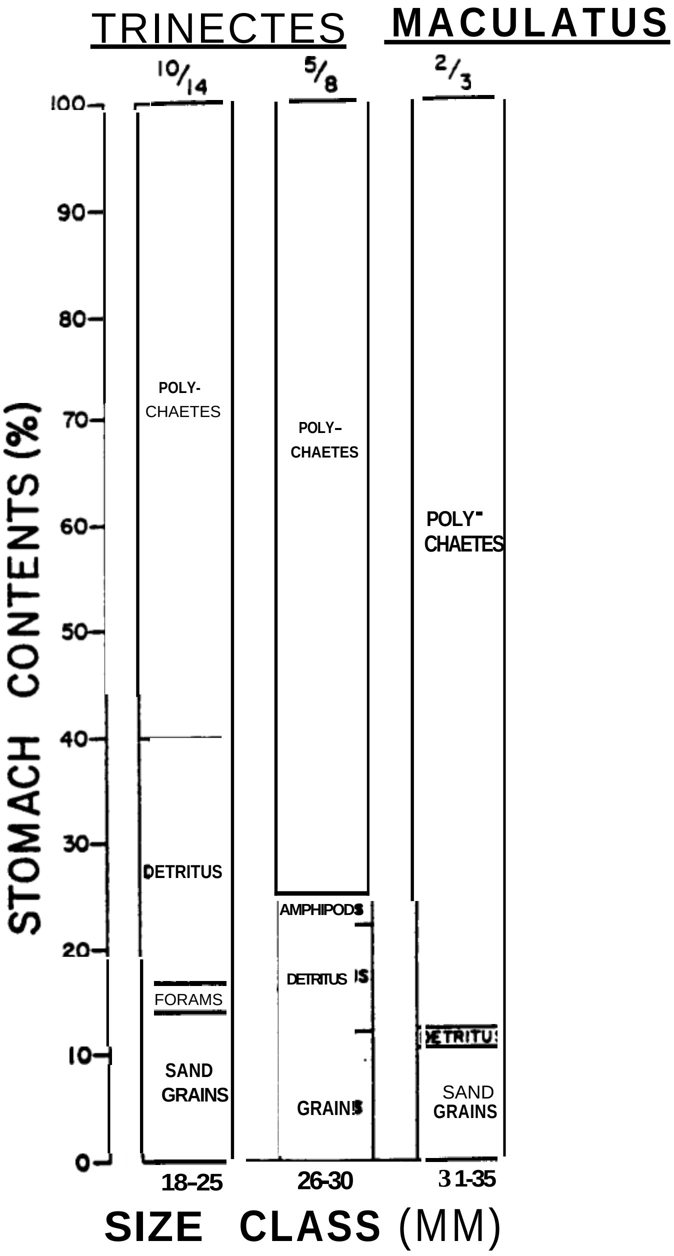

Figure 3-35. Stomach contents of juveniles of *Trinectes maculatus.* Source: Carr and Adams (1973).

Woodburn (1960) collected five hogchokers (18.7-118.5 mm). The two smallest specimens come from the bayou station during February and May (6.6-18.7 ppt); two intermediate sized fish came from Old Tampa Bay during April (19.1 ppt) and the largest specimen taken in August in Boca Ciega Bay (32.0 ppt). They suggested that larger hogchoker may prefer higher salinities than small hogchoker. However, more data are necessary to substantiate this observation.

Both lined sole and hogchoker were collected throughout Tampa Bay during FMRl's fishery independent monitoring program (FIMP 1989, 1990). Haddad et al. (1989b) reported that the hogchoker exhibited a stronger preference for fresh or oligohaline waters than the lined sole. Hogchoker also use a range of salinities during different life history stages. Larval (preflexion and flexion) hogchoker were collected in Tampa Bay near the mouth of the Little Manatee River (mesohaline (5-18 ppt) to polyhaline (>18 ppt) salinities), while post-flexion larvae and juveniles had their maximum abundances further upriver in the more freshwater reaches of the river (Peebles et al. 1992; Haddad et al. 1992; Table 3-19). Larval and juvenile hogchoker showed an increased preference for lower salinity waters with growth (Peebles et al. 1992; Fig. 3-36). Juvenile and adult hogchoker had a ubiquitous distribution throughout the Little Manatee River, but appeared to prefer waters with oligohaline to freshwater salinities (Figs. 3-37 and 3-38). While juvenile lined sole are sometimes found in very low salinities, they appear to move towards waters of increased salinity with age (Peebles, pers. comm. 1992).

Larval and juvenile lined sole were commonly collected in Tampa Bay, and only infrequently in the Little Manatee River tributary (Haddad et al. 19990; Peebles et al. 1992; Table 1). They were collected primarily at stations close to the mouth of the Little Manatee River (river kilometers 0-2.5) and were rarely reported further upstream (Haddad et al. 1990). These distribution patterns suggest that the lined sole prefers waters with mesohaline (5-18 ppt) or polyhaline (> 18 ppt) salinities in the Tampa Bay estuary.

3.11.4.2 Temperature

Both lined sole and hogchoker were collected in a wide range of water temperatures within Tampa Bay. Lined sole were collected in water temperatures ranging from 11.2-32.4^{o}C (Springer and Woodburn 1960). Hogchoker were collected in water temperatures of 14.2-30.2^{o}C during 1987-1991 (FIMP, unpublished data).

Spawning season of the lined sole may be correlated with increased water temperatures and daylength (Futch 1970). Spawning in the Tampa bay area appeared to be initiated when water temperatures reached approximately 20°C, and maximum larval abundance appeared to occur in June and July (Futch 1970).

Very little information was available to describe thermal tolerance limits of these species. Houde (1974) reported that it was difficult to rear lined sole larvae at temperatures near 32^{o}C, and he proposed that this temperature was near the upper lethal limit for this species. No lined sole or hogchoker mortalities have been reported in cold water fish kills in Tampa Bay (Rinckey and Saloman 1964; Gilmore et al. 1978). However, given the benthic nature of these species, mortalities resulting from cold water

Table 3-19. Catch data and distributional statistics for lined sole and hogchoker collected near the Little Manatee River. P = plankton net, S = seine, n = number of fish caught, f = collection frequency, S_D = density weighted mean salinity of capture, r_8 = the sign and probability value from Spearman's rank correlation between median fish length and salinity at capture; Location of A_{max} = location of highest mean fish abundance. Source: Peebles et al. (1992).

Taxon	**Stage**	**Gear Type**	**n**	**f**	S_D	r_8	A_{max}
Solidae							
Achirus Jineatus (lined sole)	preflexion	P	49	24	24.7	NS	BAY
	flexion	P	83	36	23.3	NS	BAY
	postflexion	P	202	69	21.2	p=0.03	BAY
	juvenile	P	30	19	6.0	NS	0.0
	juvenile/ adult	S	38	19	17.3	NS	0.0
Trinectes maculatus (hogchoker)	preflexion	P	210	56	19.3	NS	0.0
	flexion	P	185	65	16.8	p<0.001	0.0
	postflexion	P	433	101	10.4	p<0.001	3.8
	juvenile	P	231	69	1.7	p=0.04	10.3
	juvenile/ adult	S	21630	440	0.4	p<0.001	15.5

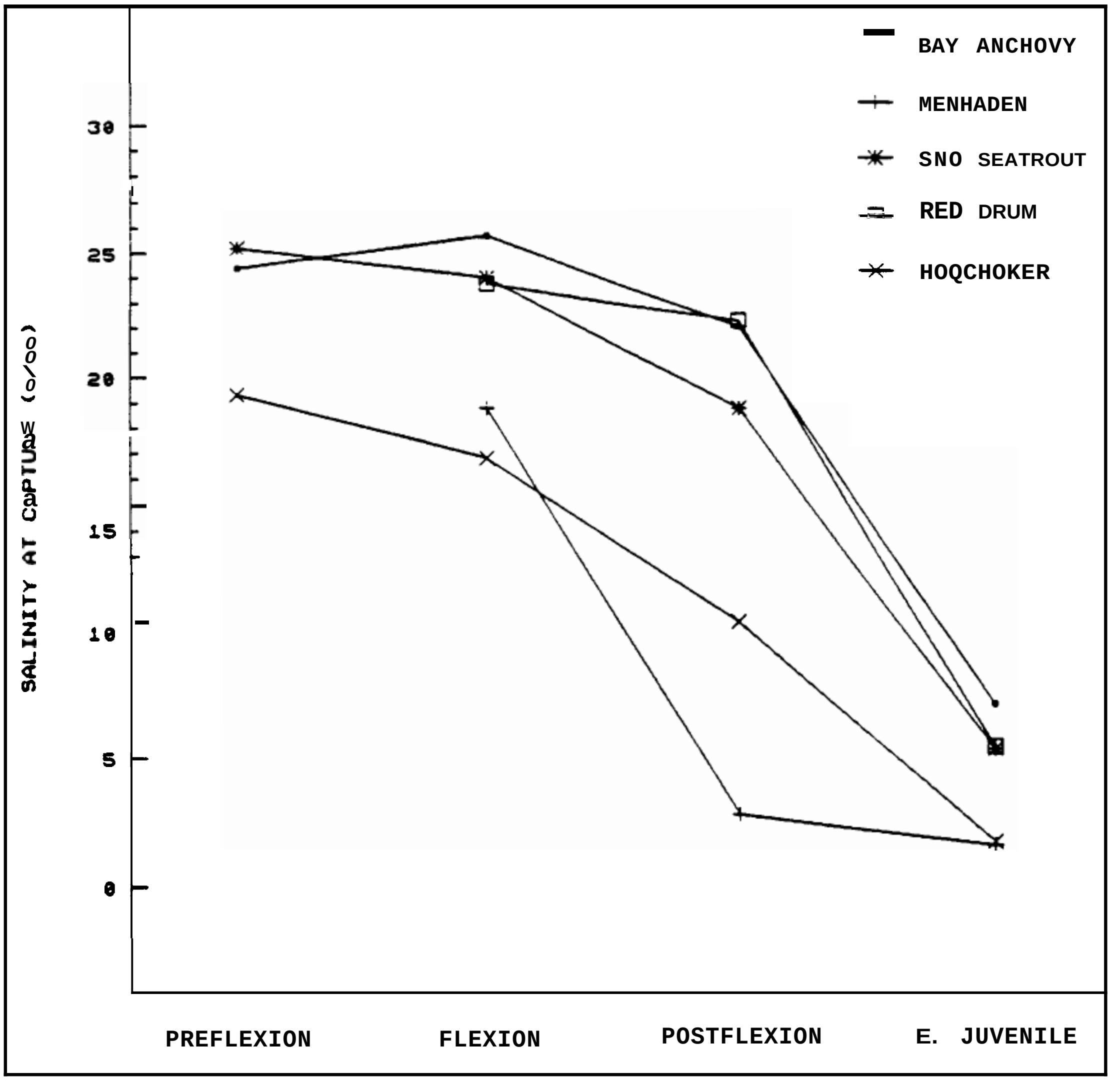

Figure 3-36. **Examples of decrease in mean salinity at capture with progressing development. Source:** Peebles et al. **(1992).**

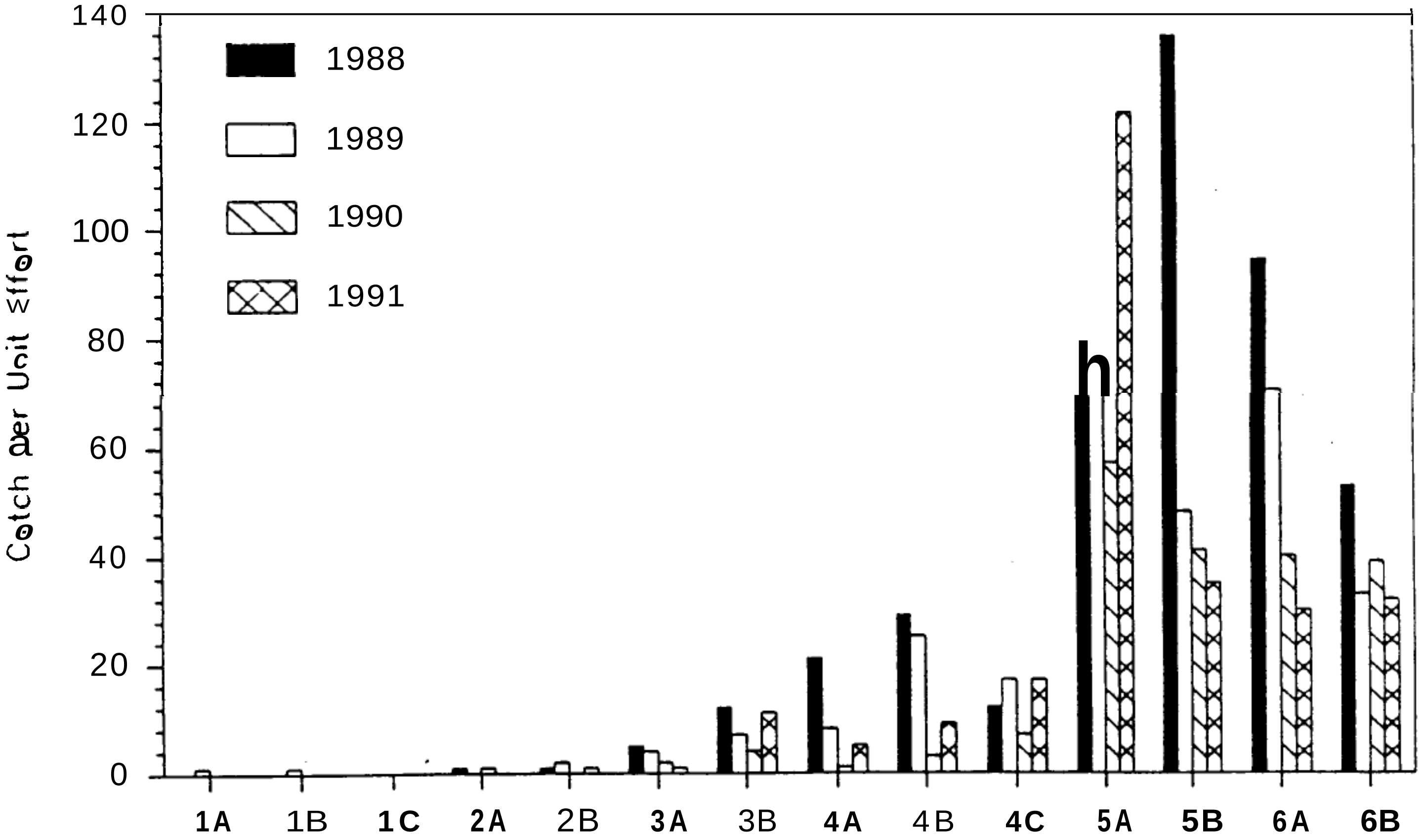

Figure 3-37. CPUE (# fish/100m^2) for juvenile and adult hogchoker in the Little Manatee River. Station 1 = *0;* station 2 = rkm 2.5; station 3 = rkm 5.8; station 4 = rkm; station 5 = rkm 15.5; station 6 = rkm 16.4. Source = Haddad et al. 1992.

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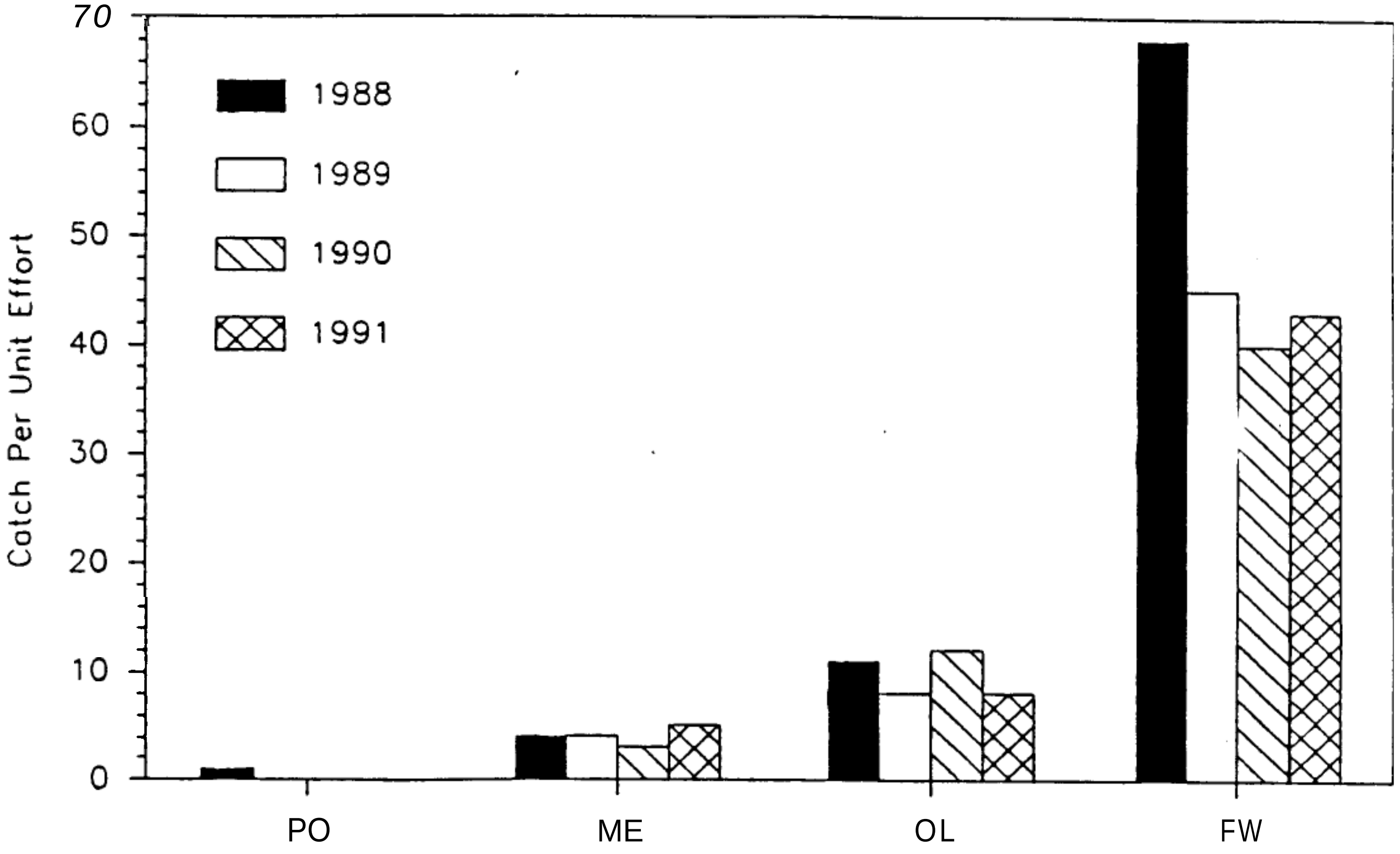


Figure 3-38. CPUE (# fish/100m^2) for juvenile and adult hogchoker by surface salinity in the Little Manatee River. PO = polyhaline (> 18 ppt); ME = mesohaline (5-18 ppt); OL = oligohaline (0.5-5 ppt); and FW = freshwater (0-0.5 ppt). Source: Haddad et al. 1992.

temperatures may not be obvious. These studies suggest that water temperature does not appear to be a critical water quality requirement affecting the abundance or distribution of these species in the Tampa Bay estuary.

3.11.4.3 Water Depth

Both hogchoker and lined sole were collected in shallow (< 2 m) seagrass and riverine stations in the Tampa Bay estuary (Springer and Woodburn 1960; Haddad et al. 1992; Peebles et al. 1992; FIMP 1989, 1990). Their distribution in deeper waters has not been adequately described.

3.11.4.4 Structural habitat

Both species were collected throughout the Tampa Bay estuary during Florida Fisheries Independent Monitoring Program (FIMP 1989, 1990). However, very little information is currently available to document their specific structural habitat preferences. A more extensive analysis of the FIMP database and other fishery independent studies might provide this information.

Hogchoker were collected on vegetated and non-vegetated bottoms throughout the Tampa Bay estuary (FIMP 1989, 1990). They were very abundant in the Little Manatee River on non-vegetated bottoms characterized by leaf litter and other detritus. Lined sole are commonly collected in seagrass areas near Cockroach Bay and the Little Manatee River (Haddad et al. 1990, 1992). Their abundance in non-vegetated areas relative to vegetated areas have not been adequately described.

Environmental requirements of lined sole and hogchoker are summarized in Tables 3-20 and 3-21.

Table 3-20. General and preferred ranges and upper and lower tolerance limits for environmental requirements of lined sole. Letters in parentheses indicate life stage. S = spawning, E = egg, L = larval, J = juvenile, A = adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				11.2-32.4 (J,A)	Springer and Woodburn 1960
	20 (S)				Futch 1970
			32 (L)		Houde 1974
Salinity (ppt)				4-35 (J,A)	Springer and Woodburn 1960
	>5 (E,L,J,A)				FIMP unpubl. data; FIMP 1989, 1990; Haddad et al. 1992; Peebles et al. 1992
Dissolved Oxygen (mg/l)					
Depth (m)	<2 (J,A)				Haddad et al. 1992
					FIMP unpubl. data
Substrate				vegetated-non-vegetated (J,A)	Haddad et al. 1990, 1992

Table 3-21. General and preferred ranges and upper and lower tolerance limits for environmental requirements of hogchoker. Letters in parentheses indicate life stage. **S**=spawning, **E**=egg, L=larval, J=juvenile, **A**=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				14.2-30.2 **(J,A)**	Futch 1970b; Houde 1974; FIMP unpubl. data
Salinity (ppt)	**>5** (L)				Springer and Woodburn 1960
	0-18 (J)			0-36 **(J,A)**	FIMP unpubl. data
Dissolved Oxygen (mg/l)					
Depth (m)				**all depths (J,A)**	Haddad **et al**. 1992; FIMP 1989, 1990; Coastal Env. Ser. 1992
Substrate				vegetated - non-vegetated **(J,A)**	Haddad **et al**. 1990, 1992; FIMP 1989, 1990
	non-vegetated **(J,A)**				Haddad **et al.** 1990, 1992; FIMP 1989, 1990

4.0 SHELLFISH

4.1 BLUE CRAB *(Callinectes sapidus)*

4.1.1 INTRODUCTION

The blue crab is economically valuable and ecologically important to the Tampa Bay estuary. It constitutes the fourth largest commercial food fishery in Florida; total annual west coast landings were 5.5 million pounds in 1991 (Steele, pers. comm. 1992). Commercial blue crab landings from the Tampa Bay area contribute about 3.6% of Florida west coast landings (Steele and Perry 1990). A substantial recreational fishery also exists in the bay; however, it is difficult to place a monetary value on its importance because recreational landings data do not exist. Ecologically, the blue crab is important to Tampa Bay because of its high abundance, omnivorous eating habits, and role as prey for upper-level carnivores. The blue crab is a vital link in the estuarine food wed as a scavenger-predator (Steele and Perry 1990).

Commercial blue crab landings data have been used in other estuaries as a rough indicator of the health of the fishery. In Florida, a general decline in blue crab production has occurred since 1960. This decline, coupled with an increase in fishing effort and a concurrent decrease in catch per unit effort (CPUE), may indicate that the blue crab fishery in Florida is overcapitalized in both labor and material (Steele and Perry 1990). Overcapitalization in the blue crab fishery appears to be a problem throughout the state; however, loss of nursery and molting habitat is also a major concern in Tampa Bay (Steele pers. comm. 1992).

The importance of commercial and recreational blue crab fisheries in the Gulf of Mexico and throughout the U.S. is reflected in the large amount of basic and applied research conducted with this species. For the Gulf States, life history and fishery characterizations for the blue crab have been described for the St Johns River, FL (Tagatz 1968a, b); Florida (Oesterling 1976), Alabama (Tatum 1979), Mississippi (Perry 1975), Louisiana (Jaworski 1972, 1982; Adkins 1982; Robert and Thompson 1982), and Texas (More 1969). The U.S. Fish and Wildlife Service has published a "Species Profile" for the blue crab within the Gulf of Mexico (Perry and McIlwain 1986). For Tampa Bay specifically, however, only limited life history information for the blue crab is available.

4.1 .1.1 Distribution

According to Williams (1974), the blue crab ranges throughout the western Atlantic from Nova Scotia to northern Argentina, including Bermuda and the Antilles. Other distributional records include Denmark, the Netherlands and adjacent North Sea, northwest and southwest France, Golfo di Genova in the northern Adriatic Sea, the Aegean Sea, the western Black sea, the eastern Mediterranean, and Lake Hamana-ko in central Japan. The blue crab is found throughout the Tampa Bay estuary; from the oligohaline tidal streams to the marine waters near the bay mouth.

4.1.1.2 Population Status and Trends

Along the west coast of Florida, annual blue crab landings (commercial) have fluctuated considerably since 1959. Production cycles span four to six years. Peak production occurred in 1970, 1977, 1981, 1984, and 1987; however, there has been a general decline in production since the 60s (Steele 1982) (Fig. 4-1).

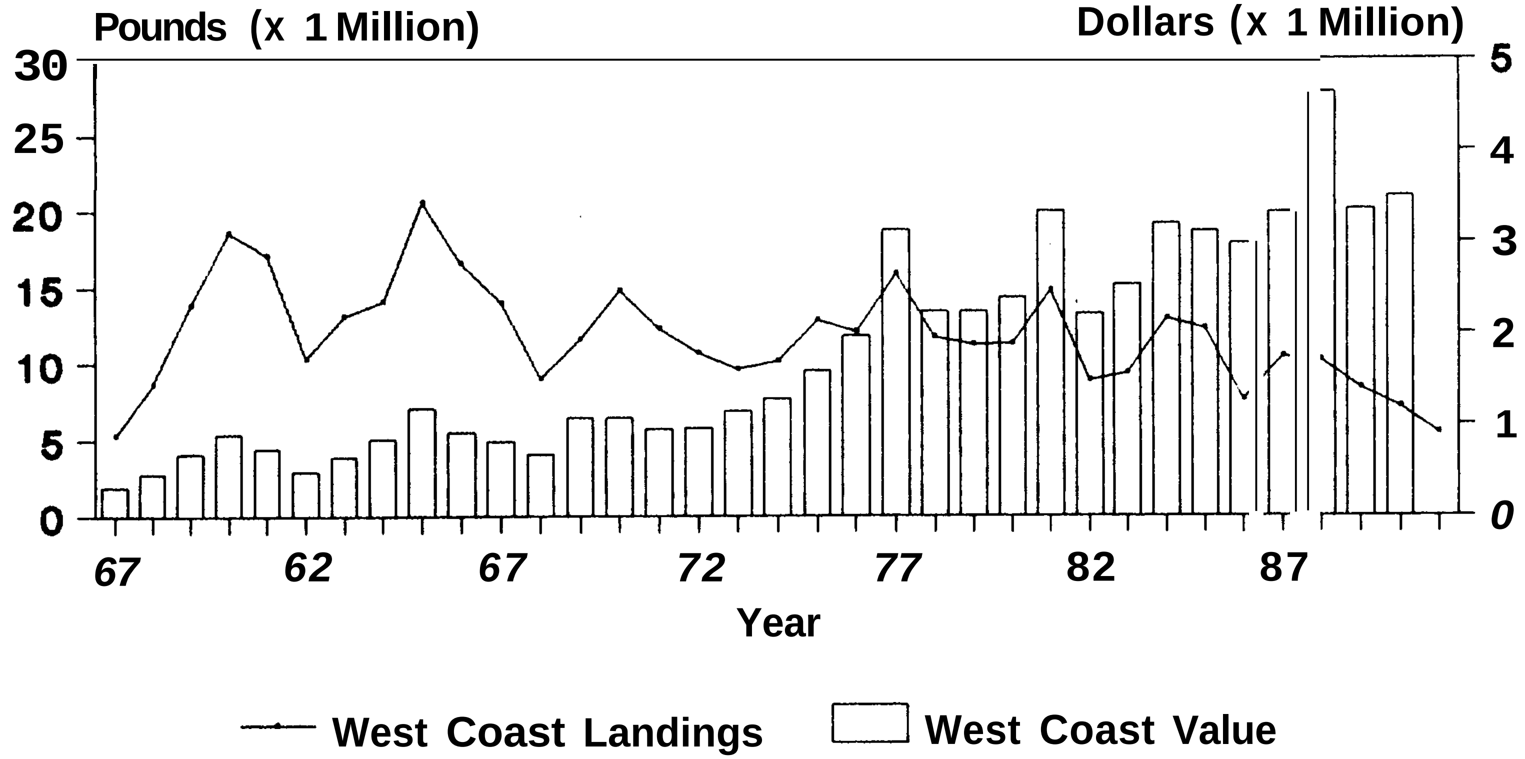
For the Tampa Bay estuary, FDNR commercial landings data for the three counties (Hillsborough, Pinellas, Manatee) surrounding Tampa Bay indicate total pounds of crabs per fishing trip have increased from 1986 to 1989 with a slight drop in 1990. These data; however, can be misleading since they are influenced by factors that cannot be accounted for such as number of traps fished per trip or where the crabs were captured (i.e., crabs may be captured in an area outside of Tampa Bay but actually sold to a crab distributor in the Bay area).

4.1.2 LIFE HISTORY

As indicated previously, information regarding early life history stages of the blue crab in Florida waters, and especially in the Tampa Bay estuary, is limited. Most studies conducted in Tampa Bay were directed more toward aspects of adult life history, including spatial and temporal distribution and reproductive biology. Consequently, the discussion of life history of blue crab larvae and small juveniles will be drawn from studies conducted in the northern Gulf of Mexico.

In Florida, the blue crab mates from March to December when water temperature exceeds 22°C (Steele 1982). As immature females approach their last juvenile molt, they migrate into male dominated, low salinity waters to molt and mate. In Tampa Bay, blue crab mating occurs in the lower salinity areas of upper Tampa Bay from midwinter through spring and September (Steele and Bert unpublished manuscript). The male couples with and inseminates the soft post molt female. Sperm will remain viable for at least one year and can be used for multiple spawnings (Tagatz 1968a).

After mating, females migrate to high salinity waters, where spawning occurs. Some females remain in the high salinity portions of lower Tampa Bay, whereas others move out into the Gulf of Mexico to spawn. Results of a tagging program conducted in Tampa Bay during 1982-83 indicated many mature female blue crabs migrate northward along the Florida west coast in the late fall-early spring (Steele 1991). Oesterling (1976) postulated that the females were moving northward to a supposed spawning site south of the Apalachicola River drainage system in northwest Florida. In addition to reproductive



1957-1986 - NMFS data;
1987-1991 - DNR Trip Tickets
No value data 1991

Figure 4-1. Florida West Coast blue crab landings and value , 1957-1991. (Source: FDNR, Florida Marine research Institute 1992) condition, Steele (1991) suggested these migrations may result in part from responses to environmental or physiological stimuli such as temperature and food availability. Further, the migration may be a behavior pattern that had adaptive significance at some earlier time during the blue crab's evolutionary history.

Females that mate late in the fall usually delay spawning until the following spring, when water temperatures are warmer (Van Engle 1958). In Tampa Bay, peak spawning occurs in high salinity areas near the mouth of the bay during March and April, with a smaller secondary peak occurring in September (Steele and Bert unpublished manuscript). Fertilized eggs are extruded and attached to the female's abdominal appendages, where they are carried for two to three weeks until hatching (Van Engle 1958).

Ovigerous females can carry 750,000 to 8,000,000 eggs (Prager et al. 1990) and are termed "sponge" crabs or "berried" females. The eggs are yellow when first extruded. As development proceeds, the eggs change from yellow-orange to brown-black, and finally to black, just before hatching. Survival is usually low; many eggs do not hatch, and still fewer larvae and small crabs survive (Steele 1982). Only one ten-thousandth of 1% (0.000001) of viable eggs survive to become adults; the remainder perish from fungal infections, predation, or excessively high or low temperatures or salinities (Van Engle 1958).

The larvae, called zoeae, usually are found offshore in high salinity waters (Millikin and Williams 1984). The zoeae pass through seven molt stages lasting 31 to 49 days and metamorphose into a megalops stage that lasts 6 to 20 days (Costlow and Bookout 1959). Currently, no data on the temporal or spatial distribution are available for larval or early juvenile blue crabs in Tampa Bay; however, Tagatz (1968a) found zoeae and megalopae in the St. Johns River, Florida during all months between April and October; most zoeae occurred in high salinity waters near the coast. Perry and Stuck (1982a) found similar results in that early stage ***Callinectes*** zoeae were present in Mississippi coastal waters in the spring, summer, and fall. Based on data from Tagatz (1968a) and Perry and Stuck (1982a), it appears that development through the late zoeal stages takes place in offshore waters, where larvae are subject to currents and can be transported considerable distances.

Recruitment of blue crabs to Gulf estuaries occurs during the megalopal stage (More 1969; King 1971; Perry 1975; Perry and Stuck 1982b). Perry and Stuck (1982b) reported that large catches of blue crab megalopae in August and September usually were followed by an increased catch of juvenile crabs (10 to 19.9 mm) in October or November in Mississippi estuaries. In Tampa Bay, megalopae probably settle out in seagrass beds near the mouth of the bay (Steele pers. comm. 1992).

First juvenile crab stage is the next molt following the megalopal stage. Juvenile blue crabs show wide seasonal and areal distribution in Gulf estuaries (Perry and McIlwain 1986). Livingston et al. (1976) found maximum numbers of blue crabs in Apalachicola Bay in winter and summer and observed that an almost "continuous succession" of juvenile crabs entered the sampling area during the year. As juvenile blue crabs develop in Tampa Bay, they continue to migrate up the estuary and undergo further growth and development.

Blue crab growth rate is dependent on water temperature, salinity, and food' availability (Leffler 1972; Holland et al. 1971; Winget et al. 1976). First stage juvenile crabs start at about 2 mm carapace width (CW) (Costlow and Bookout 1959). Male crabs reach sexual maturity in about one year, at approximately 89 mm CW (Gray and Newcombe 1938b). Size at sexual maturity for females is highly variable; they are usually mature after 18 to 20 molts, regardless of size (Van Engle 1958). Most blue crabs in St. Johns River reach harvestable size (> 127 mm CW) one year after hatching, but some may survive to four years of age (Tagatz 1968a). In Tampa Bay, Steele and Bert (unpublished manuscript) noted that most females had reached sexual maturity at 130-139 mm CW.

4.1.3 ECOLOGICAL ROLE

Blue crabs perform a variety of ecosystem functions and play a major role in energy transfer within estuaries (Van Den Avyle and Fowler 1984).

4.1.3.1 Predators

Predators of the blue crab include numerous species of fish, mammals, and birds, as well as other blue crabs. During their pelagic or open water larval stages, blue crabs are vulnerable prey for many species of small fish, other plankters, and jellyfish (Van Engel 1958). Blue crab megalopae have been reported as prey items for grass shrimp and sand shrimp (Olmi 1990), as well as larger blue crabs and fish. Cannibalism among various sizes of blue crabs has been documented in several studies (Darnell 1958; Tagatz 1968a; Laughlin 1982). Juvenile and adult blue crabs are prey for several commercially and recreationally important species of fish, such as spotted seatrout, red drum, sheepshead, and black drum. Twenty-two fish species have been identified as blue crab predators (Steele and Perry 1990) (Table 4-1), and many of these species are present in Tampa Bay. The primary mammalian predator is the raccoon. Avian predators include clapper rail, great blue heron, and American and hooded merganser (Darnell 1959; Bateman 1965; Day et al. 1973).

4.1.3.2 Diet

The food items a blue crab consumes depend on the size of the crab, habitat, and time of year (Van Heukelem 1991; Laughlin 1982). Information concerning food requirements of blue crab larvae in natural waters is limited. Van Heukelem (1991) suggested blue crab larvae probably eat rotifers, worm larvae, and copepod nauplii. After blue crab larvae molt a few times, adult copepods may be a main food item. Blue crab larvae maintained in the laboratory have been reared on rotifers, polychaete worm larvae,

Table 4-1. Fish predators of the blue crab. (Source: Steele and Perry 1990)

Species	Gunter (1945)	Darnell (1958)	Fontenot and Rogillio (1970)	Overstreet and Heard (1978a)	Overstreet and Heard (1978b)	Overstreet (Unpub. data, GCRL)	Heard (Unpub. data, GCRL)
Aplodinotus grunniens		X					
Archosargus probatocephalus	X	X	X			X	
Arius felis	X	X					
Bagre marinus	X						
Bairdiella chrysoura		X					
Caranx hippos						X	
Carcharhinus leucas							X
Cynoscion arenarius						X	
Cynoscion nebulosus	X		X			X	
Dasyatis americanus							X
Dasyatis sabina							X
Dasyatis sayi							X
Ictalurus furcatus		X					
Lagodon rhomboides		X					
Lepisosteus oculatus		X					

Table 4-1. Continued

Species	**Gunter (1945)**	**Darnell (1958)**	**Fontenot and Rogillio (1970)**	**Overstreet and Heard (1978a)**	**Overstreet and Heard (1978b)**	**Overstreet (Unpub. data, GCRL)**	**Heard (Unpub. data, GCRL)**
Lepisosteus spatula		X					
Lobotes surinamensis	X						
Micropogonias undulatus		X	X	X			
Micropterus salmoides		X					
Morone interrupta		X					
Opsanus beta							X
Paralichthys lethostigma		X				X	
Pogonias cromis	X		X			X	
Rachycentrum canadum						X	
Sciaenops ocellatus	X	X	X		X		
Sphyrna tiburo	X						

photosynthetic dinoflagellates, brine shrimp, and sea urchin eggs (Van Heukelem 1991; Sandoz and Rogers 1944). Juvenile and adult blue crabs feed on bivalves, crabs and other crustaceans, small fishes, annelids, plant matter, and detritus (Darnell 1958, Tagatz 1968a; Laughlin 1982; Alexander 1986). In Apalachicola Bay, Florida, Laughlin (1982), found differences in diet between size groups of blue crabs; juveniles under 31 mm CW fed primarily on bivalves, plant material, detritus and ostracods while blue crabs 31 to 60 mm CW fed on fish, gastropods and xanthid crabs. Crabs greater than 60 mm CW fed on fish, bivalves, xanthid crabs and other blue crabs.

In the lower Chesapeake Bay, blue crab feeding behavior may be associated with habitat; crabs in a marsh creek only fed during certain tidal cycles, whereas crabs in a seagrass bed fed continuously (Ryer 1987). Lipcius and Hines (1986) determined crab predation rates were higher in mud than in sand, and functional responses of crabs differed between the two substrates.

Time of year is also important in determining blue crab diet. Laughlin (1982) indicated that all crabs are opportunistic and prey upon whatever food items are available at the time. The diet of the blue crab changes as different prey items become vulnerable throughout the year; e.g., after spring spawning, early juvenile fish become available.

Van Heukelem (1991) points out that because the diet of the blue crab is essentially wide-ranged, it has many competitors. Changes in abundance of these competitors due to factors such as overfishing and habitat destruction ultimately affect abundance and distribution of blue crab populations.

4.1.4 CONTAMINANTS

Blue crabs are susceptible to many types of contamination, several of which have been found in Tampa Bay sediments. According to a NOAA Status and Trends Study (Long et al., 1991), sediments in parts of Tampa Bay were high in concentrations of mirex, dieldrin, DDT, PCBs, and heavy metals (silver, copper, and lead) relative to other Florida Gulf coast sites. Currently, NOAA's Bioeffects Program is conducting a study to determine if blue crabs are bioaccumulating contaminants in certain areas of Tampa Bay and to evaluate relationships between contaminant concentrations and adverse effects. Studies conducted on blue crabs from other coastal areas have documented their toxicity and bioaccumulation rates for various contaminants. The U.S. Environmental Protection Agency (Mayer 1987) has determined the acute toxicity of several chemical contaminants to blue crabs, several of which are found in Tampa Bay (Table 4-2).

In addition to Table 4-2, Bookhout and Costlow (1975) determined that mirex was toxic to blue crab larvae at 1.0 and 10.0 μg/L after five days of exposure. This contaminant was also found to be toxic to adult blue crabs at concentrations less than 0.53 μg/L during a 28-day continuous exposure (Tagatz et al. 1975). PCBs (Polychlorinated biphenyls) have been found in high concentrations in blue crab muscle and hepatopancreas by several studies (Eisenberg and Topping 1984; Marcus and Mathews

1987; Hale 1988). DDT was found to cause mortality in blue crabs at concentrations of greater than 0.005 mg/L (Lowe 1965).

4.1.5 ENVIRONMENTAL REQUIREMENTS

Steele and Perry (1990) reported that blue crab year-class strength is determined by larval recruitment and estuarine survivorship of juveniles. The influence of density-independent factors such as temperature and salinity appear to dominate larval mortality, whereas density-dependent factors such as estuarine carrying capacity are more influential during the juvenile crab stages. Interaction of these factors determines blue crab stock abundance. In general, studies have been directed at determining the physical and structural habitat requirements of juvenile blue crabs. Perry and McIlwain (1986) proposed that more research should be directed at identifying factors that affect the initial movement of larvae and postlarvae toward the estuary. Although transport mechanisms associated with blue crab larvae are poorly understood in the Gulf of Mexico, several theories have been developed relating larval transport to the Loop Current, coastal boundary layers, or coastal weather fronts; however, the physical and behavioral mechanisms that allow for seaward and subsequent shoreward transport of blue crab larvae in the Gulf are unknown (Steele and Perry 1990).

Van Engle (1982) suggested that temperature, salinity, and substrate are the primary factors affecting growth, survival, and distribution of blue crabs in the Chesapeake Bay. Variations in pollutants, predation, disease, and food availability also play a major role in influencing blue crab survival (Perry and McIlwain 1986). Although it is difficult to associate specific parameters with blue crab survival due to possible synergistic effects, several studies have been successful for various parameters and life stages.

4.1.5.1 Temperature

Water temperature influences survival, molting frequency, and growth of blue crabs, but optimal temperatures for specific life stages vary with other environmental parameters such as salinity (Zein-Eldin and Griffith 1966; Hughes et al. 1972; Winget et al. 1976). Because of this interaction between temperature and salinity, many optimal temperatures are presented with different salinities.

In order for blue crab eggs to hatch properly, prezoeal stage hatching must occur between 19° and 29°C and salinities of 23 to 33 ppt to ensure survival (Sandoz and Rogers 1944). Laboratory studies suggested that once blue crab larvae hatch, the optimal temperature and salinity combination for zoeal development was 25°C and 30 ppt (Costlow and Bookhout 1959; Sulkin and Epifanio 1975; Bookhout et al. 1976). Williams (1965) found that larvae reared at less than 21°C did not develop past the first zoeal stage and did not progress past the third zoeal stage when reared at 3O°C or higher. Development progressed normally at 25°C when salinity was between 20.1 ppt and 31.1 ppt.

Costlow (1967) determined that optimal temperature and salinity for the megalopal stage of laboratory-reared blue crabs is 25°C and 30 ppt. He also found that survival of megalopae exceeded 70% at 20°C, 25°C, and 30°C when salinity was above 10 ppt, but never exceeded 50% at 15°C.

Several studies have identified temperature and salinity tolerance limits of juvenile and adult blue crabs. Tagatz (1969) determined the 48-hour temperature tolerance limits of adult and juvenile blue crabs from the St. Johns River, Florida. Blue crabs were acclimated for 21 days to four temperatures and two salinity regimes. Juvenile and adult female crabs were less tolerant to temperature extremes at a salinity of 7 ppt compared to adult male crabs. For crabs acclimated at 30°C and 34 ppt, the upper temperature limit was 39°C, and the lower limit was 4.6 to 4.9°C. Crabs acclimated at 30°C and 7 ppt had upper temperature limits of 37°C and lower limits of 5.3 to 6.0°C. At the lowest acclimation temperature of 6°C, crabs had a low temperature tolerance limit of less than 0°C. In addition, crabs acclimated at 6°C at both high and low salinities had upper tolerance limits of 33°C and 31.5°C, respectively.

In a laboratory study, Leffler (1972) grew juvenile blue crabs at various temperatures and found that mortality was directly proportional to temperatures between 13 and 34°C. Holland et al. (1971) indicated that juvenile crab mortality increased at temperatures above 30°C and established an upper incipient lethal temperature for juvenile blue crabs at 33°C. Van Heukelem and Sulkin (1990) observed in laboratory experiments that locomotor activity of juvenile blue crabs ceased when water temperature fell to 5.5°C; however, when temperatures were elevated to 12.5°C nine days later, crabs became active again.

Steele and Bert (unpublished manuscript) conducted a two-year blue crab trapping study in Tampa Bay and found a relationship between blue crab abundance and temperature and salinity. Relative abundance of male crabs was positively correlated with temperature and negatively correlated with salinity, whereas, abundance of female crabs was negatively correlated with temperature and positively correlated with salinity. At cooler temperatures (<20°C) higher proportions of males were found in lower salinities, but at higher temperatures (>21°C) high proportions of males were collected from all salinities (>16 ppt). Temperature and salinity interactively affected the percentage of molting crabs of each sex. Molting males were collected at nearly all salinities when temperatures exceeded 16°C. Relatively high percentages of females molted at all salinities in the 26 to 30°C range and at 11 to 15 ppt in the 16 to 25°C range.

In general, frequency of blue crab molting increases as water temperatures increase (Tagatz 1968b; Leffler 1972; Winget et al. 1976); however, in Tampa Bay, the frequency of molting for both sexes appears to be inversely related to summer water temperatures (Steele and Bert unpublished manuscript). The authors suggest that midsummer water temperatures in shallow-subtropical estuaries such as Tampa Bay may be too warm to accommodate the physiological and hormonal stresses associated with growth and reproduction.

4.1.5.2 Salinity

The interactive effects of salinity and temperature on various life history stages of blue crabs have already been discussed. Additional information on salinity requirements is presented below.

Newcombe (1945) reported salinities of 22 to 28 ppt are necessary for normal hatching of eggs and development of zoeae. Larvae may hatch prematurely and die in the prezoeal stage if salinity is too low (Van Engel 1958). In contrast, specific salinity levels are not critical for the survival of postlarval blue crabs (Odum 1953; Costlow 1967; Adkins 1972; Palmer 1974). Holland et al. (1971) found that salinities within the range of 2 to 21 ppt had little effect on growth and survival of juvenile blue crabs. Juvenile blue crabs are found over a broad range of salinities; however, Perry and McIlwain (1986) stated that juvenile blue crabs in Gulf of Mexico estuaries are most abundant in low to intermediate salinities characteristic of middle and upper estuarine water.

4.1.5.3 Dissolved Oxygen

Blue crabs generally can avoid waters that contain low levels of dissolved oxygen (DO) (Van Heukelem 1991), and in some cases will actually leave the waters to avoid the hypoxic water conditions. This mass movement of blue crabs out of hypoxic waters has been documented in the Chesapeake Bay, and Alabama, and has occurred recently along the northwest coast of Florida (Steele pers. comm. 1992). This phenomenon has not been documented in Tampa Bay; however, blue crab mortality caused by low DO water is a problem in certain shallow water areas of Tampa Bay during the summer months (Steele pers. comm. 1992). Blue crabs usually cannot escape fishermen's traps placed in these shallow, low DO waters and typically'die. Trap death due to anoxia has been reported as a serious problem in many areas such as Mobile Bay, Alabama, where low DO water covered approximately 44% of the Bay in the summer of 1971, and fishermen reported up to 75% mortality of their trap catch (Tatum 1982). Carpenter and Cargo (1957) found DO levels of about 32% to 36% saturation is lethal to blue crabs that cannot escape traps. In contrast, a recent study conducted by De fur et al. (1990) found blue crabs exposed to 35% to 39% DO saturated water for up to 25 days displayed less than 20% mortality. Lowery and Tate (1986) exposed crabs to hypoxic water (< 0.5 ppm) and found they became moribund after two hours and died if not returned to water with normal DO levels within 10 to 15 minutes.

4.1.5.4 Structural Habitat

Habitat becomes an important factor in blue crab survival once megalopae enter the estuary. As megalopae move into the estuary and continue to develop, molting promotes settlement (Lipcius et al. 1990). The primary settlement habitat of blue crab megalopae appears to be seagrass or vegetated bottom (Orth and Van Montfrans 1987; Van Montfrans et al. 1990). Several studies have shown that seagrass beds and vegetated areas are important nursery habitats for megalopae and juvenile blue crabs in the Chesapeake Bay (Heck and Orth 1980; Heck and Thoman 1984; Orth and van Montfrans 1987). The association between vegetated habitat and high densities of megalopae and juvenile blue crabs may result from food availability and reduced predation rates (Shirley et al. 1990).

Adult blue crabs in the Chesapeake Bay occupy seagrass meadows and other habitats, such as unvegetated sediments, marsh creeks, and macroalgae (Heck and Thoman 1984; Kennish et al. 1984; Williams 1984). These habitats are used as sources of food, shelter, and refuge from predation (Orth et al. 1984; Williams 1984; Hines et al. 1987). Wilson et al. (1990) found that vegetation provided the best cover from predation; mean rates of predation were 9% in sea lettuce, 20% in eel grass, and 40% in unvegetated controls and marsh creeks.

Although it has been determined that vegetated areas serve as nursery grounds for juvenile blue crabs and a refuges for adult crabs, several estuaries in the Gulf of Mexico that are devoid of bottom vegetation support large blue crab populations (e.g., Mississippi Sound and Mobile Bay, Alabama). In these estuaries, juvenile blue crab distribution and abundance are associated with soft, mud sediments (More 1969; Holland et al. 1971; Adkins 1972; Perry 1975; Livingston et al. 1976; Perry and Stuck 1982b).

In Tampa Bay, limited information is available on blue crab larval distribution and abundance; however, megalopae moving into the estuary most likely settle out in the seagrass beds located in the lower bay (Steele pers. comm. 1992). As indicated earlier, blue crabs do not require seagrass beds for settlement, and may settle on other types of structure (Perry pers. comm. 1992). Habitat preference of small juvenile blue crabs in Tampa Bay is being investigated and appears that they probably use seagrass beds in the same manner as those in the Chesapeake Bay (Steele pers. comm. 1992).

Steele and Bert (unpublished manuscript) documented habitat partitioning displayed by adult and large juvenile blue crabs in Tampa Bay and concluded that salinity rather than substrate type or benthic vegetation may be a major factor influencing blue crab abundance and distribution. They found a greater number of blue crabs inhabiting upper Tampa Bay relative to the lower bay. In addition, male blue crabs were larger and more abundant in the lower salinity waters of the upper bay, whereas subadult males and large females were found occupying the mid and lower portion of the bay. It was also suggested that one station in the lower bay may serve as a refuge for less fit or older crabs. The most remarkable aspect of size-specific site utilization in Tampa Bay was its constancy over time; temporal and spatial variation in abundance, size, and sex ratio was similar at each station between the two sampling years (1981-1983).

Environmental requirements of blue crab are summarized in Table 4-3.

Table 4-2. Toxicity to Blue Crabs[a]

Chemical	Temperature (°C)	Salinity (ppt)	Duration (h)	Flow[b]	Test	Concentration
Aldrin (insecticide)	28	21	48	FT	EC_{50}	23 μgL^{-1}[c]
Antimycin A (piscicide)	25	29	48	FT	EC_{50}	> 100 μgL^{-1}[c]
Azinphos-Methyl (insecticide)	27	27	48	FT	EC_{50}	320 μgL^{-1}[c]
Carbaryl (insecticide)	30	28	48	FT	EC_{50}	320 μgL^{-1}[c]
Chlordane (insecticide)	29	23	48	FT	EC_{50}	260 μgL^{-1}[c]
Chlordecone (Kepone) (insecticide)	19	20	96	FT	LC_{50}	>210 μgL^{-1}[d]
Chlorpyrifos (insecticide)	17	20	48	FT	EC_{50}	5.2 μgL^{-1}[c]
2,4-D Propylene Gl Butyl Ether Ester (herbicide)	24	29	48	S	EC_{50}	2,800 μgL^{-1}[c]
Dieldrin (insecticide)	18	26	48	FT	EC_{50}	240 μgL^{-1}[c]
Endosulfan (insecticide)	30	24	48	FT	EC_{50}	19 μgL^{-1}[c]
Endrin (insecticide)	11	16	48	FT	EC_{50}	15 μgL^{-1}[c]
Fenthion (insecticide)	28	25	48	FT	EC_{50}	2.3 μgL^{-1}[c]
Hepachlor (insecticide)	17	27	48	FT	EC_{50}	68 μgL^{-1}[c]
Malathion (insecticide)	30	25	48	FT	EC_{50}	> 1,000 μgL^{-1}[c]
Methoxychlor (insecticide)	31	27	48	FT	EC_{50}	320 μgL^{-1}[c]
Mirex (insecticide)	31	24	48	FT	EC_{50}	> 2,000 μgL^{-1}[c]
Naled (insecticide)	28	25	48	FT	EC_{50}	220 μgL^{-1}[c]
Ozone (water sterilant)	25	7.4	96	S	LC_{50}	0.26 mgL^{-1}[d]
Toxaphene (insecticide)	19	27	48	FT	EC_{50}	180 μgL^{-1}[c]

b

c

d

Table 4-3. General and preferred ranges and upper and lower tolerance limits for environmental requirements of blue crab. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)	19-29 (E)				Sandoz and Rogers 1944
	25 (L, at **30** ppt)				Costlow and Bookhout 1959; Sulkin and Epifanio 1975; Bookhout et al. 1976
		21 (L)	**30** (L)		Williams 1965
			33 (J)		Holland et al. 1971
		5.5 (J)			Van Heukelen and Sulkin **1990**
				variable depending on salinity and sex	Steele and Bert (unpubl. manuscript)
	23-33 (E)				Sandoz and Rogers **1944**
	30 (**L** at 25°C)				Costlow and Bookhout **1959;** Sulkin and Epifaunio **1975;** Bookhout et al. **1976**
Salinity (ppt)	22-28(E)				Newcombe **1945**
				2-21 (J)	Holland et al. **1971**
				variable depending on age and sex	Steele and Bert (unpubl. ms)
D.O. (mg/l)		<0.5 (J,A)			Lowery and Tate **1986**
Substrate	seagrass (L)				Lipcius et al. **1990;** Steele pers. comm. **1992**
				seagrass, unvegetated, macroalaae (A)	Steele pers. comm. **1992;** Heck and Thoman **1984;** Williams **1984**

4.2 PINK SHRIMP *(Penaeus duorarum)*

4.2.1 INTRODUCTION

The pink shrimp is economically and ecologically important to the state of Florida as well as to Tampa Bay. Commercial shrimping, which includes pink shrimp and two other penaeids, is Florida's number one fishery (FDNR 1989). In Tampa Bay, the pink shrimp also supports a sizeable bait-shrimp fishery, along with a small recreational dipnet fishery. Pink shrimp are a vital link in the marine food web, directly affecting the quality of the fisheries in Tampa Bay. Many economically valuable fish species such as spotted seatrout and snook may heavily utilize pink shrimp as a food source (Tabb 1961; Marshall 1958).

4.2.1.1 Distribution

The pink shrimp is found from the lower Chesapeake Bay to the Florida Keys and along the Gulf of Mexico. The largest populations of pink shrimp are off southwestern Florida and in the southeast portion of the Gulf of Campeche (Perez Farfante 1969).

4.2.1.2 Status and Trends

During the mid-1950s and 196Os, Tampa Bay bait shrimpers brought in some 50 million shrimp; however, by 1981, only three million were caught in the bay (Saloman 1965; Weintz 1987). During this time, fishing pressure was considered low, and averaged about seven boats per day. A bait-shrimp fishery still exists in the bay; however, fishing pressure is high (at least 10 times the effort in the 1960s) and the catch is limited (Chiandusse, pers. comm. 1992). According to the Florida Marine Research Institute's commercial landings data, pink shrimp landings (non-bait) remained relatively stable at two to three thousand pound per trip from 1986 to 1990 with a slight decline occurring during 1991 (Brown, pers. comm. 1992). Declines in seagrass and other aquatic vegetation, which serves as primary nursery habitat for juvenile pink shrimp, have been linked with declines in shrimp abundance (Saloman 1965; Haddad 1989).

4.2.2 LIFE HISTORY

4.2.2.1 Reproduction

When pink shrimp become sexually mature (males at 74 mm total length (TL) and females at 85 mm TL) (Eldred et al. 1961), they emigrate from the estuary to deeper offshore waters to spawn (Williams 1955). Near Tampa Bay, Eldred et al. (1965) reported spawning occurred near the mouth of the bay (4-10 m deep) to areas 40 miles offshore

(28-42 m). Rising water temperatures trigger spawning activity (Eldred et al. 1965). Most spawning occurs between 19.6 and 30.6°C (Jones et al. 1970). Spawning usually occurs in Tampa Bay and offshore areas from April through November, with peaks from April through September (Eldred et al. 1961, 1965). The female pink shrimp usually carries between 42,000 to 624,000 eggs (Martosubroto 1974). Eggs are approximately 0.23 to 0.33 mm in diameter (Eldred et al. 1965), and demersal (Ewald 1965).

When the eggs hatch, the larvae proceed through five naupliar stages, three protozoeal stages, and two to five mysis stages before reaching a postlarval stage (Ewald 1965). Under laboratory conditions, the duration of the larval development was 15 days in waters of 26°C and 21 days at 21°C (Ewald 1965). The pelagic larvae are carried by currents into the estuaries. Jones et al. (1970) and Kennedy and Barber (1981) reported that larvae may use tidal currents to enter the estuarine nursery grounds. At stations located near the mouth of Tampa Bay, Eldred et al. (1965) found large numbers of postlarvae during May through September with a peak in July. Postlarvae enter estuarine and coastal bay nursery areas at 8 mm TL (Copeland and Truitt 1966) and become benthic at about 10 mm TL (Costello and Allen 1970).

Pink shrimp remain in nursery areas approximately two to six months (Costello and Allen 1966) and become available to the bait-shrimp fishery at about six weeks of age (47 mm TL) (Saloman 1965). Pink shrimp become sexually mature at approximately 9 or 10 weeks of age (Bielsa et al. 1983). The sex ratio of pink shrimp can vary according to differences in size of shrimp, geographic area and season (Eldred 1961). In Tampa Bay, Eldred (1961) observed that females were more common than males among shrimp less than **55** mm TL, males were more prevalent among shrimp between **55** and **85** mm TL, and females were again more numerous than males at sizes greater than **85** mm TL. Saloman (1965) determined that females in Tampa Bay outnumbered males by an narrow margin and were generally larger in average size. Females in Tampa Bay were more numerous than males from April to July and from September to December, whereas males were predominant only in January, and for the remaining months the sex ratio was close to 1:1 (Eldred et al. 1961).

After pink shrimp grow to a size of about 95 to 100 mm TL, they begin to emigrate from the estuarine nursery areas to offshore waters (Joyce 1965). In Tampa Bay, Eldred et al. (1961) found that large adult shrimp (85 to 140 mm TL) migrated out of the bay from April through July. Other authors suggest emigration may occur year-round with a major peak in the fall and a minor one in the spring (Huff and Cobb 1979; Kennedy and Barber 1981). It appears that pink shrimp have an average life span of approximately 83 weeks (Kutkuhn 1966); however, Eldred et al. (1961) reported pink shrimp may reach or exceed two years of age.

4.2.2.2 Growth

Numerous factors, such as developmental stage, water temperature, sex of the shrimp, and food availability, influence the growth rate of pink shrimp (Iversen and Jones

1961). From the nauplius stage to the postlarval stage, Dobkin (1961) observed a rapid growth rate of 3.72 mm (0.38 to 4.1 mm TL) in a period of two to three weeks. In Tampa Bay, Saloman (1968) observed that juveniles grew an average of 1.2 mm carapace length (CL) per month from August through May. For subadult and adult shrimp, monthly growth rates have been determined to range from 0 to 22 mm TL (Costello and Allen 1960; Iversen and Idyll 1960; Iversen and Jones 1961; Costello 1963; Knight 1966)

4.2.3 ECOLOGICAL ROLE

4.2.3.1 Diet

Pink shrimp are omnivorous animals and eat a variety of plant and animal material. Larval shrimp about 10 mm TL feed almost exclusively on filamentous blue-green algae and diatoms (Flint 1956). Juveniles and adults are generally bottom feeders, especially in shallow inshore areas where bottom vegetation is present (Eldred et al. 1961). In Tampa Bay, juvenile and adult pink shrimp feed on dinoflagellates, foraminiferans, nematodes, polychaetes, ostracods, copepods, mysids, isopods, amphipods, coridean shrimps, caridean eggs, and mollusks. Sand, debris, algae, diatoms, seagrass, and fish scales have also been found in shrimp digestive tracts (Eldred et al. 1961). Pink shrimp generally feed more in the summer than the winter and Williams (1955) reported that stomachs were nearly always empty in colder months.

4.2.3.2 Predators

Pink shrimp are prey to numerous fish, bird, and other wildlife species. They are a major food source for many fish species, several of which, including spotted seatrout and snook, are economically important to Tampa Bay (Tabb 1961). Other fish species present in the Tampa Bay vicinity that are major predators of pink shrimp include; mangrove snapper ***Lutjanus griseus,*** red grouper ***Epinephelus morio,*** black grouper ***Mycferopercabonaci,*** and king mackerel ***Scomberomorus cavalla*** (Costello and Allen 1962, 1970). Bottle-nosed dolphins, which are common in Tampa Bay, also have been identified as predators of the pink shrimp (Miyazaki et al. 1973). Pink shrimp are preyed on by many different bird wading and diving bird species (Bent 1926) and various reptile species (Costello and Allen 1970).

4.2.4 CONTAMINANTS

Limited information is available on the effects of contaminants on pink shrimp. The studies available primarily consider the impacts of petroleum and other oil products on penaeid shrimp; all studies indicated adverse effects ranging from total mortality to bioaccumulation (Couch 1978; Soto et al. 1981; Botello et al 1981).

4.2.5 ENVIRONMENTAL REQUIREMENTS

4.2.5.1 Temperature

In Tampa Bay, pink shrimp have been captured in water temperatures ranging from 10 to 35.5°C (Eldred et al. 1961,1965; Saloman 1965, 1968). Cook and Murphy (1969) determined that optimum temperatures for larval growth and survival of penaeid shrimp were 24 to 32°C. Reynolds and Casterlin (1979) determined from laboratory experiments that juvenile and adult pink shrimp preferred water temperatures of 22 to 36°C at night and 17 to 38°C during the day. Regarding a lower temperature limit, cold water mortality in the natural environment has not been reported for this species; however, at temperatures of 13.3°C, pink shrimp become narcotized. They may recover fully once water temperature increases (Eldred et al. 1961). Cold water mortality has been observed in a bait tank, and Eldred et al. (1961) suggests the minimum survival temperature for pink shrimp in Florida waters is around 12°C.

Similar to cold water mortality, no records exist of pink shrimp death caused by high water temperatures (Costello and Allen 1970). The pink shrimp behavior of burying into the substrates possibly serves as a protection for the shrimp during extremely high summer temperatures as well as very low winter temperatures (Eldred et al. 1961).

4.2.5.2 Salinity

Pink shrimp are found in a variety of salinities depending on life stage. Eldred et al. (1965) collected larvae and postlarvae near Tampa Bay at salinities about 37 ppt. Tabb et al. (1962) found postlarvae at salinities ranging from 12 to 43 ppt in Florida Bay. Williams and Deubler (1968) found postlarvae at salinities as low as 0.50 ppt. Juvenile and adult shrimp size and salinity is positively correlated (Williams 1955; Gunter et al. 1964; Tabb et al. 1962; Saloman 1962); however, Saloman (1968) found more large shrimp at lower salinities than at higher salinities in Tampa Bay. In Florida Bay, Tabb et al. (1962) captured juveniles in salinities that ranged from 5 to 47 ppt and adults in salinities from 25 to 45 ppt.

The synergistic effects of temperature and salinity together can impose strict environmental restraints on shrimp populations (Bielsa et al. 1983). Williams (1960) found that as temperature decreases, shrimp are not as tolerant to salinity changes; however, at high salinities they were better able to tolerate low water temperatures.

4.2.5.3 Dissolved Oxygen

Limited information is available concerning the dissolved oxygen requirements of pink shrimp. Most studies that relate dissolved oxygen to shrimp document oxygen consumption rates rather than specific tolerance levels or requirements.

4.2.5.4 Water Current

Water currents are important to the pink shrimp, primarily for distribution. Postlarvae depend on tidal currents to transport them into the estuarine nursery areas (Hughes 1969). Juveniles utilize ebbing tidal current for emigration out of the estuary to offshore waters (Burkenroad 1949; Hughes 1969).

4.2.5.5 Light

Light is an important factor in controlling activity of pink shrimp (Bielsa et al. 1983). Pink shrimp are considered nocturnal (Eldred et al. 1961; Fuss and Ogren 1966). In laboratory studies, adult pink shrimp burrowed in the presence of solar light, and when light intensity diminished to less than 0.01 1 lumens per m^2 they became active (Fuss and Ogren 1966). However, pink shrimp 55 to 105 mm TL were shown to display positive phototaxis to 3.229 lumens per m^2 in the laboratory (Aaron and Wisby 1964). Fuss and Ogren (1966) showed a difference in reaction to light in relation to shrimp size. These authors observed that large pink shrimp were more sensitive to light than small ones. Eldred et al. (1961) reported that in Tampa Bay, larger pink shrimp were caught in greater numbers at night than during the day.

4.2.5.6 Substrate

Pink shrimp select different bottom types depending on their life stage. Subadults prefer shell sand and loose peat (Williams 1958); whereas adults prefer calcareous sediments or hard sand bottoms (Hildebrand 1955). Offshore of Tampa Bay, Huff and Cobb (1979) found that adult pink shrimp abundance was high in sand, sand-shell, or exposed limestone habitats.

In the estuaries, juvenile pink shrimp require cover for protection against predators and to provide an adequate food supply (Williams 1955). In Tampa Bay, salt marshes, mangroves, and especially seagrasses provide both protection and a food source for pink shrimp (Thayer et al. 1978). The importance of aquatic vegetation cover such as seagrasses to the survival of pink shrimp juveniles has been recognized by the following authors: Williams (1955), Woodburn et al. (1957), deSylva (1954), Allen and Inglis (1958), Hutton et al. (1956), Hoese (1960), Phillips (1960), Woodburn (1959), Hildebrand (1955), Tabb et al. (1962), and Turner (1977). In addition, it has been documented that inshore shrimp fisheries do not exist where seagrasses are rare or absent (Hoese and Jones 1963). In Tampa Bay and other Florida waters, the loss of seagrass has been related to the decline of the bait-shrimp fishery (Saloman 1965; Weintz 1987).

4.2.5.7 Water Depth

Adults are most abundant at depths of 11 to 65 m (Huff and Cobb 1979), but also have been captured at depths of 110 m (The U.S. Bureau of Commercial Fisheries 1961).

Environmental requirements of pink shrimp are summarized in Table 4-4.

4.3 AMERICAN OYSTER *(Crassostrea virginica)*

4.3.1 INTRODUCTION

The American oyster is an important commercial and recreational species along the Atlantic and Gulf of Mexico coasts. In Tampa Bay, oyster abundance has gradually declined from the early 1900s to a point where the commercial oyster fishery in the bay is essentially non-existent. The importance of the oyster to Tampa Bay, besides supporting a small recreational fishery, is primarily ecological. Tampa Bay oyster reefs, whether dead or alive, provide structural habitat which is important for the survival of many commercially and recreationally valuable fish and invertebrate species. Over 40 macrofaunal species or groups live in oyster beds (Bahr and Lanier 1981). and the total number of species in an oyster community may exceed 300 (Wells 1961). Many of the species which use these reefs are sources of food for economically important species. A recent management plan for the oyster fishery in the Gulf of Mexico provides detailed information on the life history, distribution, and habitat requirements of this species (Berrigan et al 1991).

4.3.1.1 Distribution

The American oyster is found in nearshore, estuarine ecosystems from the Gulf of St. Lawrence, Canada to the Yucatan Peninsula, Mexico, including the Gulf of Mexico (Galtsoff 1964; Abbott 1974). This species has been introduced to the Pacific coast of North America, the Hawaiian Islands, Japan, Australia, and the United Kingdom (Ahmed 1975), but apparently with limited success. Major oyster fisheries are found in the northern Gulf of Mexico, the Chesapeake Bay, and Long Island Sound.

In Tampa Bay, both live and dead oyster reefs exist along the shallow inter- and sub-tidal regions of the bay (Fig. 4-2). Several of these areas are privately leased through the State of Florida.

4.3.1.2 Status and Trends

Fossil shell deposits and early fishery records substantiate the fact that oysters were at one time very abundant in Tampa Bay (Cooke 1945; Dawson 1953). Commercial oysters were in high abundance throughout the region during the late 1800s, at which time Hillsborough County was second in commercial production of oysters in Florida with oyster meat production levels approaching 500,000 pounds annually (Dawson 1953). From 1902 to 1962 the fishery gradually declined to 5,000 or fewer pounds per year. After 1962, oyster landings increased temporarily, probably due to the use of clutch

Table 4-4. General and preferred ranges and upper and lower tolerance limits for environmental requirements of pink shrimp. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				10-35.5	Eldred et al. 1961, 1965; Saloman 1965, 1968
	24-32 (L)				Cook and Murphy 1969
	22-36 (J,A) night				Reynolds and Casterlin 1979
	17-38 (J,A) day				Reynolds and Casterlin 1979
		12 (J,A)			Eldred et al. 1961
Salinity (ppt)				to 37 (L)	Eldred et al. 1965
				12-43 post (L)	Tabb et al. 1962
				as low as 0.5	Williams and Deubler 1968
				5-47 (J)	Tabb et al. 1962
				25-45 (A)	Tabb et al. 1962
Dissolved Oxygen (mg/l)					
Depth (m)	11-63(A)				Huff and Cobb 1979
				to 110 m	U.S. Bureau of Commercial Fisheries
	<2 (J)				Haddad 1989; Thayer et al. 1978

Table 4-4. Continued

	Preferred	Lower Limit	Upper Limit	Range	Reference
Substrate	shell sand and loose peat				Williams 1958
	calcareous sediment, hard sand (A)				Hildebrand 1955
	seagrass (J)			salt marshes, mangrove, seagrass (J)	Thayer et al. 1978
	seagrasses (J)				numerous authors (see text)

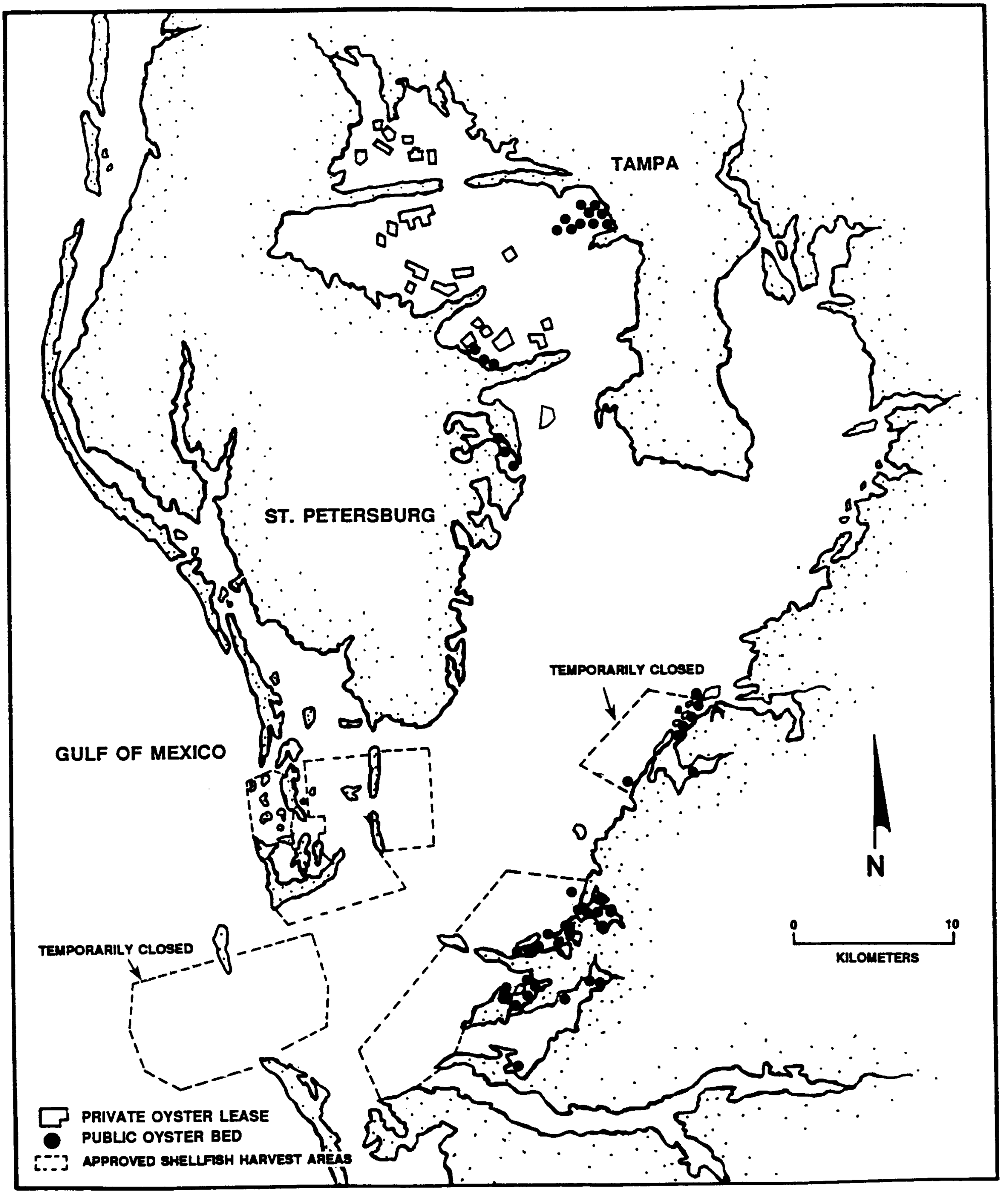


Figure 4-2. Distribution of American oyster in the Tampa Bay estuary. Summarized from Kunneke and Palik 1984

(oyster shells) on leased oyster grounds covering more than 1,000 acres in Old Tampa Bay (Finucane and Campbell 1968). However, after the 1960s, the oyster fishery began to decline again until it finally collapsed during the 1980s. In 1982 only 132 pounds of oysters were landed in the Tampa Bay area (Snell 1984) and from 1986 to 1990 only one commercial trip that produced 18 pounds was recorded in the Florida Marine Research Institute's commercial landings data base (Brown pers. comm. 1992). Although oyster abundance has significantly decreased in Tampa Bay mainly because of anthropogenic activities, the decline in commercial landings over the past 10 years also is more a function of shellfish closures (Berrigan, pers. comm. 1992). Only a few areas in lower Tampa Bay are open to shellfish harvest (Fig. 4-21, the rest of ,the bay has been restricted because of high fecal coliform levels.

The oyster decline in Tampa Bay can be attributed to several factors which are primarily associated with effects of continued development in and around the bay area (Wolfe and Drew 1990). Dredge and fill activities, stormwater runoff, and effluent discharges increase turbidity and sedimentation in the aquatic environment and silt may settle out and cover oyster spat, inhibiting normal development. This soft muddy habitat that results from sedimentation is undesirable for oyster spat settling. Stormwater runoff and point source discharges can also transport contaminants into oyster reef areas causing sublethal and lethal stress in oyster populations. In addition to these factors, certain parasites and diseases have been attributed to the oyster decline in Tampa Bay (Dawson 1953).

4.3.2 LIFE HISTORY

4.3.2.1 Reproduction

Oyster populations in the Gulf of Mexico generally begin spawning when water temperatures are above 2O°C. When temperatures climb above 25°C, mass spawning occurs (Schleselman 1955). In Tampa Bay, oysters probably spawn during all months except when temperatures reach extreme highs or lows (Berrigan, pers. comm. 1992; Quick and Mackin 1971). Major spawning peaks in Tampa Bay probably occur in the spring and fall with the fall spawn being the more successful (Berrigan, pers. comm. 1992). Male oysters usually spawn first, and their release of sperm and pheromones trigger other males and females to begin spawning (Cake 1983). In the northern Gulf of Mexico, mature oysters spawn numerous times and this is probably true for oysters in Tampa Bay (Berrigan, pers. comm. 1992).

Female oysters can produce 23 to 86 million eggs per spawning and a positive relationship exists between size of the female and number of eggs produced (Davis and Chanley 1955); however, other factors such as level of maturation and ambient water conditions affect female fecundity. Once the eggs are released and fertilized, they become demersal and sink to the bottom where they are dispersed by currents and internal waves. (Glatsoff 1964). Egg development depends on water temperature. Generally, the embryo becomes a trochophore larva after several hours (Cake 1983).

4.3.2.2 Larval Development

Once the egg hatches, the trochophore larvae remains in the water column and develops, for at least two to three weeks (Bahr and Lanier 1981). The trochophore larvae develops into the first of a series of free-swimming veliger larvae (Cake 1983). The first stage veliger is called the straight-hinge or D-stage larva and is characterized by a ring of locomotory cilia (velum) and two shells (prodissoconch). This stage then develops into a pediveliger larva (prodissoconch II) which is characterized by a pronounced umbone. The pediveliger larva has two eyes and an elongate foot, but can still swim (Andrews 1979). The foot functions as a tactile mechanism, exploring the substrate before settlement. Once a preferable bottom habitat is located, liquid "cement" is extruded from a pore in the foot and the left valve become fixed in place (Kennedy 1991). After attachment, larvae lose the velum and foot and are classified as spat. Spat that set during the first three days after metamorphosis may grow faster than those setting later (Losee 1979). Metamorphosis can be delayed if suitable substrate is unavailable. In laboratory experiments, Coon et al. (1990) kept Pacific oyster larvae from setting for 30 days. Anthropogenic inputs may possibly cause premature setting or inhibit the process by interfering with the larvae's biochemistry (Kennedy 1991). The time from spawning to setting varies depending upon geographic location and factors such as water temperature and condition. In Galveston Bay, Texas, setting occurred two months after spawning (Hopkins 1931).

During larval development, veligers are passively transported via water currents and generally remain within the estuary. However, they are capable of moving vertically to utilize tidal currents to remain in areas of preferable water quality (Carriker 1951; Galtsoff 1964; Wood and Hargis 1971). This type of dispersal ensures the species' survival in favorable areas of an estuary, even if traditional reef areas become unacceptable because of adverse environmental conditions (Berrigan et al. 1991).

In the northern Gulf of Mexico, American oysters can develop into sexually mature adults in a period of 4 to 12 weeks after settlement. Spawning by young of the year (YOY) and production of two generations of oysters could occur in one year (Menzel 1951; Hayes and Menzel 1981). However, YOY spawning does not contribute significantly to the year class because of the low number of eggs produced (Hayes and Menzel 1981). Two generation production within one year may also occur in Tampa Bay (Berrigan, pers. comm. 1992).

Adult American oysters are dioecious, but can exhibit protandrous hermaphroditism (Bahr and Lanier 1981). Usually, young oysters are predominately males, but over subsequent breeding seasons can become females (Coe 1934; Galtsoff 1961, 1964). In the James River Estuary, Virginia, sex ratios changed from 90% males at one year of age to 80% females in older oysters (Andrews 1979).

4.3.2.3 Age and Growth

In general, American oysters grow fastest during their early months of life (Bahr 1976). Berrigan (1990) estimated that oysters in Apalachicola Bay, Florida grew about 44.2 mm during their first year with 25% of the population exhibiting growth of 49.9 mm/year. Similarly, Butler (1954) reported that oysters in the Gulf region grow about 50 mm/year. Growth slows considerably in large, older oysters when metabolic reserves are needed to maintain reproductive activities and soft tissues (Stenzel 1971). Gulf of Mexico oysters can survive for 10 or more years provided they are not subjected to stresses associated with harvesting, predation, diseases, or burial (caused by adverse sedimentation rates) (Cake 1983).

4.3.3 ECOLOGICAL ROLE

4.3.3.1 Diet

Oyster larvae are planktivorous and prefer to prey on small naked flagellates, especially chrysophytes (Guillard 1957). However, Davis and Calabrese (1964) reported that at high water temperatures (greater than 27°C) naked flagellates are not as abundant, and oyster larvae tend to feed on chlorophytes. Bacteria are apparently not eaten by oyster larvae (Davis 1953).

The diet of adult oysters is not clearly understood; however, the gills of the adult oyster have been reported to retain diatoms, dinoflagellates, and graphite particles from 2 to 4 μm in length (Bahr and Lanier 1981). Bacteria are sometimes consumed, presumably because they are attached to detritus (Stanley and Sellers 1986).

The adult oyster filters a substantial amount of water. The volume of water filtered per hour is about 1500 times the volume of the oyster's body (Loosanoff and Nomejko 1946). The filtration rate is independent of the available food supply, the stage of tide, or the time of day (Stanley and Sellers 1986). Newell (1988) hypothesized that the decline of the filter feeding oyster assemblages in the Chesapeake Bay may have been an important factor in the apparent shift to microbial food webs and increased zooplankton in the bay.

4.3.3.2 Predators

All developmental stages of the American oyster are vulnerable to various types of predators, many of which are found in Tampa Bay. As a result of predation and occasional disease, oysters in Tampa Bay generally suffer high natural mortality (Berrigan, pers. comm. 1992; Quick and Mackin 1971). Oyster larvae are prey to several types of planktivores (e.g., ctenophores) and benthic carnivores (e.g., sea anemones) as well as other filter feeding invertebrates (Kennedy 1990). Newly settled spat are eaten by species such as the carnivorous flatworms (e.g., ***Stylochus ellipticus)*** whereas older spat and first year oysters are preyed on by numerous species including crabs (e.g., blue and stone crabs), fish (e.g., black drum, skates, and rays), conchs (e.g., ***Melongena corona),*** whelks (e.g., ***Busycon contrarium)*** and southern oyster drills (Marshall 1954; Menzel and Nicky 1958; Chapman 1959; Schessellman 1955; Hopkins 1955, Butler 1954; Gunter 1955; Menzel et al. 1966).

American oysters are susceptible to various diseases depending on the water quality parameters, especially temperature and salinity. In Tampa Bay, oysters may become infected with the pathogenic protozoan ***Perkinsus marinus,*** particularly when water temperatures are high during the summer (Quick and Mackin 1971). Berrigan (1990) suggested that some oyster mortality in an oyster reef restoration project in Apalachicola Bay, Florida, may have been caused by P. ***marinus.*** Quick and Mackin (1971) estimated that over 50% of Florida's adult oysters were killed by the protozoan ***P. marinus.***

An additional source of natural mortality is red tide. Outbreaks of red tide can occur in Tampa Bay several times a year and it has been determined that blooms of red tide ***(Colchlodinium heterolobatum)*** at concentrations of 500 cells/ml can kill oyster larvae (Ho and Zubkoff 1979).

4.3.4 CONTAMINANTS

Long et al. (1991) conducted a study directed at identifying status and trends of toxicants, and their potential for biological effects in Tampa Bay. Concentrations of toxicants in bivalve tissues commonly associated with adverse effects were compared with concentrations in Tampa Bay bivalves. They also compared concentrations in oysters to the proposed criteria for fish and shellfish intended to protect predatory wildlife (National Academy of Sciences 1974).

These authors found that compared to oysters from other NOAA Status and Trends sites along the Gulf Coast, those in Tampa Bay had relatively high concentrations of mirex, chlordane, Hg, and Zn. Among nationwide sampling sites, the concentrations of total chlordane, mirex, Hg, As, and Zn were relatively high in oysters from Tampa Bay.

Based upon comparisons of the chemical concentrations previously associated with biological effects and the concentrations observed in Tampa Bay, there appears to be a small to moderate potential for adverse effects among Tampa Bay biota that would be attributable to exposure to toxicants. The potential for toxic effects among resident oysters likely would be relatively low as a result of accumulation of PAHs, DDT, chlordane, dieldrin, Cu, and Pb in their tissues and moderate as a result of contamination by PCBs, Hg, and Zn (Long et al. 1991).

4.3.5 ENVIRONMENTAL REQUIREMENTS

As an estuarine resident, the American oyster is exposed to fluctuating environmental parameters such as temperature, salinity, and dissolved oxygen. Because of the oyster's tolerance to these fluctuations, it becomes difficult to precisely define their environmental requirements, especially when several are acting synergistically (Stanley and Sellers 1986). Many studies (laboratory and field) have been conducted to determine specific environmental requirements of the American oyster. Pertinent information from these studies is given below.

4.3.5.1 Temperature

Water temperature affects oyster reproduction, development, growth, and feeding activity (Kennedy 1991). In general, optimum water temperatures for growth, reproduction, and overall survival of the American oyster range from about 20 to 3O°C (Stanley and Sellers 1986). The earlier stages of oyster development appear to be more adversely affected by extreme temperature changes. Hidu et al. (1974) subjected fertilized eggs, ciliated gastrulae, and 2-day-old veliger larvae to temperatures of 30°C and higher for a period ranging from 10 minutes to 16 hours. They found that the fertilizated eggs were least resistant to the high temperatures, followed by the gastrulae and veliger larvae. MacInnes and Calabrese (1979) determined that embryos begin to develop abnormalities when temperatures decline from 20 to 15°C or increase to 35°C. Davis and Calabrese (1964) documented a maximum larval growth rate for Long Island oysters at temperatures between 30 and 32.5°C.

Adult oysters can be very tolerant of extreme temperature changes for short periods of time. During low tide, oysters have been observed to survive air temperatures below freezing or above 49°C (Galtsoff 1964). However, high temperatures (above 35°C) lasting an entire tidal cycle have been shown to cause oyster mortality in the Indian River bay, Delaware (Tinsman and Maurer 1974). In a laboratory study, Henderson (1929) determined that the critical thermal maximum for the American oyster was 48°C; however, Kennedy (1991) suggests this maximum is environmentally unrealistic and is not ecologically useful.

In Florida, Quick (1971) determined the effects of constant high temperature (35°C) on the American oyster. He found substantial mortalities did not occur at this elevated temperature; however, the oysters experienced altered gametogenesis, glycogen decreases, and tissue damage. In addition, they were unable to spawn the following year. Water temperatures of 37°C in Redfish Bay and Harbor Island, TX, have resulted in mass mortalities of oyster populations (Copeland and Horse 1966).

4.3.5.2 Salinity

Salinity is perhaps the single most important factor influencing the distribution and abundance of esturine organisms. It is particularly important with respect to oysters

(Berrigan et al. 1991). Salinity requirements for oysters vary depending on geographic location, life stage, and water quality parameters such as temperature. Wild populations of oysters in the Gulf need a proximate location to freshwater discharges such as rivers, creeks and bayous. These discharges provide nourishment while diluting the higher salinity Gulf waters. Successful oyster setting and growth are dependent on this median salinity environment where they are generally afforded protection from high salinity predators and disease (Berrigan et al 1991).

Salinity tolerance of oyster larvae is related to the salinity at which their parents spawned (Davis 1958). Larvae produced from eggs of adult oysters found in low salinity (8.7 ppt) can tolerate salinities that range from 7.5 to 22.5 ppt whereas larvae from eggs in high salinity waters (26 to 27 ppt) can tolerate 12.5 to above 35 ppt (Davis 1958). In order for oyster larvae to set and develop normally, salinities must range from 5 to 35 ppt, with optimal setting occurring between 10 and 30 ppt (Carriker 1951; Davis 1958; Calabrese and Davis 1970). In Tampa Bay, spat set in salinities that ranged from 16 to 30 ppt (Finucane and Campbell 1969).

In the Gulf of Mexico, oyster reef growth and reproduction is optimal at salinities between 10 and 30 ppt, but they can survive salinities from 5 to 40 ppt (Gunter and Geyer 1955; Butler 1954; Galtsoff 1964; Stenzel 1971). In Tampa Bay, healthy adult oysters were collected in salinities that ranged from 4.9 to 30 ppt (Dawson 1953). Extremely low salinities have been shown to inhibit gonadal maturation in oysters in Chesapeake Bay (Butler 1949) and Long Island Sound (Loosanoff 1953). Gunter (1953) reported high oyster mortality caused by spring floods in Mississippi Sound and Louisiana marshes, which lowered salinities (less than 2 ppt) for extended periods of time. However, short periods of low salinity can benefit oysters by killing off predators that cannot tolerate the low salinity water (Owen 1953; Marshall 1954).

Salinity tolerance is inversely correlated with ambient water temperature. Higher water temperatures generally reduce salinity tolerance. Oysters are tolerant of low salinity conditions at temperatures below 5°C, but can survive only a few days under the same conditions when the temperature is 15°C (Andrews 1982).

4.3.5.3 Dissolved Oxygen

Limited information exists on the dissolved oxygen (DO) requirements of the American oyster. Oysters are facilitative anaerobes and are able to tolerate hypoxic conditions (0-2 ppm) and survive brief exposures to anoxic conditions (Berrigan et al. 1991). Sparks et al. (1958) documented oysters surviving in water containing less than 1.0 mg/L of oxygen for up to five days. Widdows et al. (1989) found that oyster larvae (82 μm) can withstand anoxic waters for approximately 11 hours and that oyster spat (16 mm) can survive for about 150 hours. Kennedy (1991) noted that oyster larvae could swim vertically in the water column to avoid low DO waters.

4.3.5.4 Currents

Adequate movement of water or current velocity is required for normal development of oysters. Current is important for supplying food sources, removing feces and pseudofeces (to prevent burial), and maintaining sufficient water quality (Berrigan et al. 1991). Water flow requirements are poorly understood. For maximum feeding, current velocity must be high enough to exchange the water above the reef three times every hour (Galtsoff 1964). Veal et al. (1972) calculated that tidal flows of 156 to 200 cm/sec or higher are necessary for optimum growth of oysters in Mississippi. However, Mackenzie (1981) observed that a velocity of 150 cm/sec caused unattached oysters to be swept away in Long island Sound. In North Carolina, current velocities have been recorded at 11 to **66** cm/sec on oyster bars in Beaufort Inlet and the Newport River (Wells 1961).

4.3.5.5 pH

pH can influence oyster reproduction and development. In Long Island Sound, oysters spawned at pH 7.8 to 8.2, but not below pH 6.0 or above pH 10 (Calabrese and Davis 1969). Oyster embryos have been found to develop normally within a pH range of 6.75 to 8.75; however, at a pH above 9.0 or below 6.5, abnormal development occurs (Calabrese and Davis 1966). Larvae tolerate the same pH range as embryos but growth is optimum at pHs of 8.25 to 8.5 (Calabrese and Davis 1966).

4.3.5.6 Water Depth

Oysters occur at depths of 0.3 m above to 12 m below mean low tide (Butler 1954). Oysters were observed in depths that ranged from 0.3 m to 5 m in a survey conducted in Tampa Bay (Dawson 1953).

4.3.5.7 Substrate

In the Gulf of Mexico, oysters are found in shallow bays, mud flats, and offshore sandy bars (Butler 1954; Copeland and Hoese 1966; Menzel et al. 1966). In general, oysters prefer bottom types that are firm, such as those of shell, rock, and firm or sticky mud (Kennedy 1991). Oysters along the Gulf are most successful in shallow bays and on mud flats. They can survive on relatively dense mud that is firm enough to support their weight. Soft mud and shifting sand are the only substrates unsuitable for oyster communities (Galtsoff 1964). In Tampa Bay, oysters are found on all bottom types described above (Dawson 1953). Maximum oyster setting is on horizontal surfaces (Clime 1976) and the most suitable substrate for spat settlement is oyster shell (Kennedy 1991).

Environmental requirements of American oyster are summarized in Table **4-5.**

Table 4-5. General and preferred ranges and upper and lower tolerance limits for environmental requirements of oyster. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	**Lower Limit**	**Upper Limit**	**Range**	**Reference**
Temperature (°C)	**20-30 (A)**				Stanley and Sellers 1986
	30-32.5 (L)				Davis and Calabrese 1979
		15 (L)	35 (L)		MacInnes and Calabrese 1964
			35 (A)		Quick 1971;Tinsman and Mauer 1974
Salinity (ppt)	10-30 **(L)**			5-35 (L)	Carriker 1951, Davis 1958, Calabrese and Davis 1970
				16-30 **(L)**	Finucane and Campbell 1969
	10-30 (A)			5-40 (A)	Butler 1954, Gunter and Geyer 1955, Galtsoff 1964, Stenzel 1971
				5-30 (A)	Dawson 1953
		<2 (A)			Gunter 1953
D.O. (Mg/L)				to <1 (L,A)	Sparks et al. 1958, Widdows Et al. 1989
Currents (cm/secl	156-200+ (L,J,A)				Veal et al. 1972
				11-66 (L,J,A)	Wells 1961
			150 (L,J,A)		Mackenzie 1981
Depth (m)				0.3-5 (L,J.A)	Dawson 1973
				0.3-12 (L,J,A)	Butler 1954
pH	7.8-8.2 (S)	6 (S)	10 (S)		Calabrese and Davis 1969
	6.75-8.75 (L)	6-5 (L)	9 **(L)**		Calabrese and Davis 1966

Table 4-5. Continued

	Preferred	Lower Limit	Upper Limit	Range	Reference
Substrate	firm shell, rock (L,J,A)				Kennedy 1991
				mud flats, sandy areas hard bottoms (L,J,A)	Kennedy 1991

4.4 HARD CLAMS

Northern Quahog ***(Mercenaria mercenaria)***
Southern Quahog ***(Mercenaria campechiensis)***

4.4.1 INTRODUCTION

Currently, both the northern and southern quahog are present in varying abundance throughout Tampa Bay and support a small localized fishery that is primarily recreational. Tampa Bay was the site of several hard clam surveys to determine the abundance and potential for a commercial fishery (Sims and Stokes 1967; Godcharles and Jaap 1973). Benthic studies in Tampa Bay by the Bureau of Commercial Fisheries Biological Laboratory (currently the Florida Marine Research Institute) produced information concerning age and growth (Saloman and Taylor 1969), distribution, and habitat preference of the southern quahog (Taylor and Saloman 1968a,b, 1969, 1970; Taylor et al. 1970; Sykes and Hall, 1970). These studies, coupled with recent studies performed on hard clams in the Indian River (Hesselman et al. 1989; Jones et al. 1990; Arnold et al. 1991), provide important information about environmental requirements and critical habitats of the hard clam in Tampa Bay.

4.4.1.1 Distribution

Along the Atlantic coast, the southern quahog ranges from New Jersey (Merrill and Ropes 1967) to the St. Lucie Inlet, Florida (Godcharles and Jaap 1973), and ranges from Florida to Texas in the Gulf of Mexico (Ladd 1951). The northern quahog is found in intertidal and subtidal areas along both coasts. Its range extends from the Gulf of St. Lawrence to Texas (Belding 1912; Johnson 1934; Abbott 1954, 1974), but it is most abundant from Massachusetts to Virginia (Stanley and Dewitt 1983), and is common in waters from North Carolina to the Florida east coast (Arnold, pers. comm. 1992).

In Tampa Bay, the northern quahog is not as abundant as the southern quahog. It usually occurs in high density pockets near oyster beds and small tidal creeks. The southern quahog is abundant in sand bottom areas and is spread over a larger area in the estuary (pers. comm. Arnold, 1992). Sims and Stokes (1967) conducted a hard clam survey in Tampa Bay during 1964 and found populations were primarily located in the lower portions of the Bay. Apparently this area once supported larger clam populations than it does today. All clams collected were the southern quahog; however, they noted that fishermen had transplanted the northern quahog from the U.S. east coast into areas of Old Tampa Bay.

To enhance Florida's clam fishery, northern quahogs were introduced into the state of Florida in the early 1960s (Sims and Stokes 1967). The seeding program called "Operation Baby Clam" transplanted northern quahogs grown in Massachusetts to areas along the coast of Florida, including Tampa Bay. The remnant northern quahog populations that presently exist in Tampa Bay may be a result of this operation. The distribution of hard clams in Tampa Bay is shown in Figure 4-3.

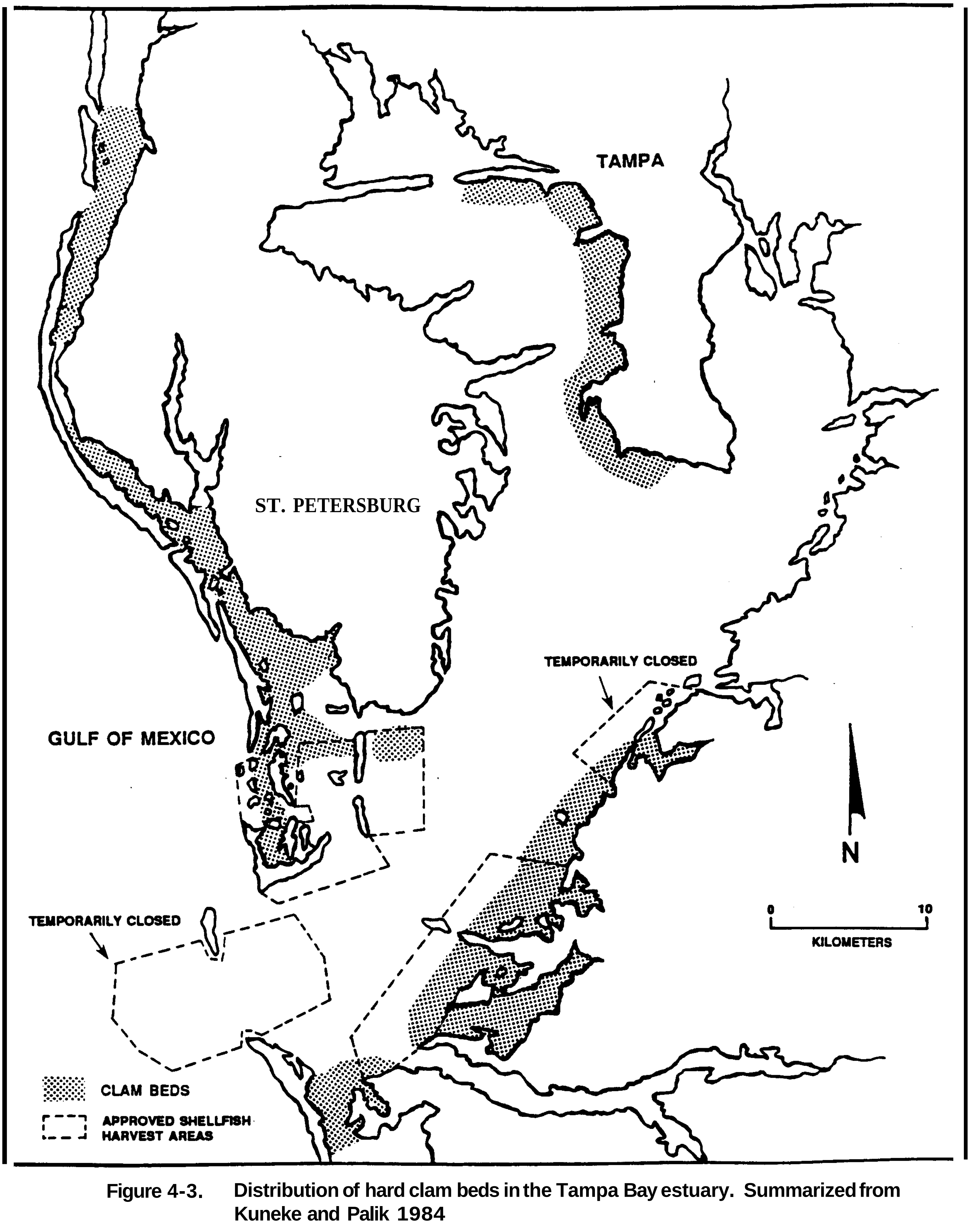


Figure 4-3. Distribution of hard clam beds in the Tampa Bay estuary. Summarized from Kuneke and Palik 1984

4.4.1.2 Population Status and Trends

A vast bed of southern quahogs existed for many years along the lower Florida, West Coast from Cape Romano southward through the Ten Thousand Islands (Schroeder 1924). This area supported a sizeable fishery from 1913 to 1947. This fishery declined during the late 1940s, and surveys have indicated no appreciable recovery (Godcharles and Jaap 1973; Arnold, pers. comm. 1992). During this time a clam fishery existed in Tampa Bay, but was described as being short-term.

A very insignificant commercial hard clam fishery still exists in the Tampa Bay estuary. The commercial clam fishery has declined over the past 10 years to become almost non-existent primarily because many of the clam beds have been restricted or closed due to poor water quality (Fig. 4-3). According to the Florida Marine Research Institute commercial landings data for the three counties surrounding Tampa Bay, pounds of hard clams landed have declined from 241 lb/trip in 1986 to less than one pound per trip during the following years (Brown, pers. comm. 1992). Since 1986, less than 20 reported trips have occurred per year, emphasizing the small size of the fishery and the effect of shellfish closures. These reported trips do not include recreational harvest which is undoubtedly much greater than commercial harvest in the Tampa Bay estuary, although no estimates of recreational harvest are available.

4.4.2 LIFE HISTORY

4.4.2.1 Spawning and Reproduction

The hard clam (both northern and southern quahog) is classified as a protandric hermaphrodite. Most individuals of the population begin adult life as males (Eversole et al. 1980; Dalton and Menzel 1983). During the subsequent winter, approximately 50% of the adult population become females (Loosanoff 1937) which results in individuals of both sexes available for reproduction. Generally, hard clam populations greater than one year of age exhibit a sex ratio of approximately 1:1 (e.g., Bricelj and Malouf 1980). Sexual maturity is primarily a function of size (Bricelj and Malouf 1980; Loosanoff 1936, 1937). Lossanoff (1936,1937) determined that northern quahogs develop functional male gonads at **6-7** mm in shell length (SL) in the first or second year of life. Oocytes may be present in the gonad at this time. After reaching approximately **30** mm SL, sexual maturity occurs and the hard clams function as either males or females (Ansell 1967).

Information relating to reproductive activity of the northern and southern quahog specifically to Tampa Bay does not exist; however, Hesselman et al. (1989) identified the reproductive cycle of a hard clam population in the Indian River located along the east-central coast of Florida. This region is near the southern distributional limit of the northern quahog (Abbott 1974). The southern quahog is found within the Indian River estuary and readily hybridizes with the northern quahog (Dillon and Manzi 1988). Tampa Bay is located at a similar latitude as Indian River and also contains northern and southern populations; therefore, reproductive activity of the hard clams between the two estuaries is probably similar (Arnold, pers. comm. 1992). In Indian River spawning occurs year round. Hesselman et al. (1989) reported that females were in spawning condition during all months and males in all months except one. However, a distinct bimodal spawning pattern was evident, with a peak occurring in the spring (February-June) and a lesser peak in the fall (September-December).

Water temperature is the determining factor in the final maturation of the gametes. When stimulated by the appropriate temperature, males release semen containing pheromones. Semen and associated pheromones are transported by water currents to the females, which then are stimulated to release eggs (Nelson and Haskin 1949). Fertilization occurs throughout the water column. The planktonic eggs are discharged by the female through an exhalent siphon. Eggs are surrounded by a thick gelatinous membrane that swells upon contact with water. This membrane causes the eggs to be buoyant so that water currents can disperse them. The sperm released by the male clam contacts the eggs and fertilization occurs. To ensure fertilization, sperm are usually much more numerous than eggs.

Northern quahog spawning is induced at temperatures of 20 to 25 °C throughout its range (Keck et al. 1975; Manzi et al. 1985). In the Indian River population, which consists of northern and southern quahogs and their reciprocal hybrids, spring spawning commenced in February as water temperature exceeded 18.5 °C and continued until June when the temperature reached 28.3 °C. The fall spawning period (September to December) corresponded to water temperatures of 29.0 to 14.5 °C (Hesselman et al. 1989). No specific information exists on the reproductive cycle of the adult southern quahog. However, in northern Florida, laboratory-spawned southern quahogs had a bimodal spawning period which was similar to those of the northern quahogs and reciprocal hybrids (Dalton and Menzel 1983). These authors concluded that the spawning cycle of northern and southern quahogs in Florida waters was identical.

The age of first reproduction of the male hard clam varies depending on factors such as water temperature and food availability. Eversole (1986) estimated that the northern quahog reaches sexual maturity at a shell length of approximately 33 mm. In Tampa Bay, male clams probably reach sexual maturity at 20 to 30 mm shell length and about a year of age (Arnold, pers. comm. 1992). Females will reach maturity about a year later at approximately 40 mm shell length. Size, not age, determines sexual maturity, so that slower growing individuals mature later than 2 years of age. Peak reproductive potential is reached at about 60 mm and larger, older hard clams gradually lose reproductive capacity and transform into a state of senility (Belding 1931).

4.4.2.2 Fecundity and Eggs

Estimates regarding hard clam fecundity are variable. Laboratory tests report that an average sized female northern quahog can produce about a million eggs per season (Davis and Chanlely 1956; Ansell 1967). Bricelj and Malouf (1980) noted female hard clams about 60 mm SL produced an average of 6.3 million eggs over one spawning season. The larger the clam, the greater number of eggs are produced (to 60 mm shell length). However, egg survival is related to egg size, not to clam size (Ansell 1967; Kraeuter et al. 1981).

4.4.2.3 Larval Development

According to Loosanoff et al. (1966), there are no substantial differences between the pattern and timing of development in the northern and southern quahogs. Both species follow the development process of blastula, gastrula, trochophore, straight-hinged (90-140 μm), umboned (140-220 μm), and pediveliger (170-230 μm) stages (Chanley and Andrews 1971; Loosanoff and Davis 1963). After fertilization, the egg reaches a two-celled stage in about 45 min. and a four-celled stage in about 90 min. At 22 °C, the trochophore stage is reached about 12 hours after fertilization (Loosanoff and Davis 1950).

The trochophore is capable of moving through water with a ciliated velum (Loosanoff and Davis 1950). The trochophore (90 x 65 μm) develops a mouth and a shell gland. Approximately 24 to 36 hours following fertilization, the shell gland finalizes secretion of the shell and the early veliger becomes a "straight hinge" form (Carriker 1961). The straight-hinged veliger form lasts 1 to 3 days. The veliger then develops into the "umboned veliger" form, which is characterized by an umbone projecting above the middle of the hinge line (Carriker 1961).

After 6 to 20 days, the umboned veliger reaches the pediveliger stage, which marks the beginning of its benthic life. The pediveliger has a foot along with the velum and can swim short distances as well as crawl along the bottom. This stage terminates when the velum is lost and the clam begins benthic existence exclusively. Upon setting, the pediveliger secretes the Dissoconch shell (Carriker 1961). Initially the larva becomes a byssal plantigrade and affixes itself to the substrate with a byssus. This stage is referred to as the spatting stage. The clam is not permanently attached by the byssus however, and can alternate between byssal attachment and crawling to find a desirable environment. This will continue for several weeks until reaching approximately 9 mm (Belding 1912; Carriker 1961).

The next developmental stage is termed the "juvenile plantigrade" stage, and is identified when the clam loses its byssus and must maintain its position beneath the sediment surface by means of its foot. Siphons are fully developed at this stage (Carriker 1961).

As the clam grows, movement decreases and the adult clam burrows deeper and remains in much the same location for the rest of its life (Belding 1911). Adult hard clams bury into the substrate, approximately 2 cm in sand and 1 cm in mud (Pratt and Campbell 1956) and have limited horizontal movement. Chestnut (1952) reported that adult clams moved up to 15 cm laterally in 38 days, and Kerswill (1941) documented movements of up to 30 cm in 2 months.

4.4.2.4 Growth

Jones et al. (1990) determined growth of northern and southern quahogs in Florida by collecting specimens from estuaries around the state, and also analyzing existing data from Boga Ceiga Bay in Tampa Bay (Saloman and Taylor 1969). The authors concluded that the northern quahog grew fastest in spring and late fall, less in winter, and slowest in summer. The southern quahog exhibited a similar pattern but apparently grew slowest in winter. The greatest growth was observed in populations of the southern quahog from Boca Ceiga Bay where the largest known living specimens of hard clams (over 150 mm) occur. Jones et al. (1990) reported that only 4% of Florida clams they collected had attained the age of 20; therefore, they concluded that both species of ***Mercenaria*** from Florida grow faster than their counterparts to the north, but do not seem to live as long.

4.4.3 ECOLOGICAL ROLE

4.4.3.1 Diet

Larval and adult hard clams are primarily phytoplankton or microalgae suspension feeders. Loosanoff and Davis (1963) observed that larval clams were capable of selective feeding similar to adults by presenting them with a mixture of algal cells and noting that the larvae selected the relatively larger cells of ***Chlamydomonas*** and rejected the cells of ***Porphyridium.*** In addition to planktonic microalgae, adult clams eat suspended detritus and its associated bacteria (DiDomenico and Iverson 1977). Tenore and Dunstan (1973) conducted clam feeding studies and determined food densities of 300 mg/l of carbon are optimal for feeding.

4.4.3.2 Predation

The primary natural control of hard clam populations is predation (Virstein 1977). The impact of predation is felt by all size classes, with even the largest clams incapable of complete escape. The clam is preyed on by fish, birds, starfish, crabs, and other mollusks. Planktonic invertebrate and vertebrate predators consume massive numbers of free-swimming clam larvae. It has been estimated that one such predator (the ctenophore ***Mnemiopsis leidyi)*** may crop 5-10% of the plankton population per day during ctenophore blooms (Kremer 1979).

Juvenile and adult hard clams fare no better than their younger counterparts, and in fact predation may increase in severity as the animal increases in size from two to 10-20 mm SL (Arnold 1984). In Florida estuaries, including Tampa Bay, decapod crustaceans account for the greatest proportion of predatory losses in hard clams (pers. comm. Arnold 1992), especially mud crabs (Landers 1954; Whetstone and Eversole 1977) and blue crabs (MacKenzie 1977; Arnold 1984). Gastropod molluscs, including ***Busycon*** spp. (Magalhaes, 1948; Peterson 1982), the drills ***Thais haemastoma*** (Butler 1953), and

Urosalpinx cinera, the moon snails ***Polinices duplicatus*** and ***Lunatia heros*** (MacKenzie 1977) are also important predators of the hard clam.

Published reports of fish predation on hard clam populations are rare, but there are indications that some degree of fish predation does occur (pers. comm. Arnold 1992). Fish such as the black drum, puffers, and flounders have been known to prey upon localized concentrations of clam seed. Skates and rays may be important predators of localized clam populations (Kraeuter and Castagna 1980; Walker and Tenore 1984). In general; however, these animals are of minor importance when compared with invertebrate predators (Carter 1968).

The sea star ***(Asterias forbesii)*** preys upon juvenile and adult hard clams, although this occurs primarily in the northern areas. Small clams are attacked by individual starfish, but clams larger that 50 mm SL are commonly attacked by an aggregation of sea stars (Doering 1981).

Several different types of shorebirds prey upon bivalve molluscs, however their influence is restricted to intertidal areas. For example, herring gulls ***(Larus argentatus)*** have been observed capturing large hard clams and dropping them over hard substrates to break them open (Schneider 1978).

4.4.3.3 Contaminants

Embryos and larval hard clams are much more susceptible to contaminants than adults. Adults often can withstand large body burdens of toxic materials, and can concentrate these substances far above ambient concentrations (Roegner and Mann 1991). These authors discuss effects of various organic and inorganic compounds. Concentrations of petroleum products in the low mg/L range are toxic to embryonic and larval clams (Roegner and Mann 1991). The hard clam is was very sensitive to waste motor oil (Byrne and Calder 1977).

Polynuclear aromatic hydrocarbons (PAH) were found to accumulate in hard clams much faster than they are depurated, giving bioaccumulation factors in the $10^3 - 10^4$ range (Bender et al. 1988).

Heavy metals including Ag, Cu, Fe, Hg, Mn, Ni, Pb and Zn were toxic to eggs and larval hard clams in the µg/L to mg/L range (Carriker 1961; Calabrese et al. 1977a, b; Calabrese et al. 1982). Metals are concentrated in hard clams at several orders of magnitude greater than the surrounding environment. Although most of these studies were conducted on northern hard clams in the Chesapeake Bay region, it is likely that hard clams in the Tampa Bay estuary are subject to and affected by similar contaminants. For example, it is known that Tampa Bay sediments contain relatively high concentrations of heavy metals such as Ag, Cu and Pb (Long et al. 1991). These metals are most likely incorporated into hard clam tissues and could potentially affect reproductive potential or survival, depending upon life stage.

4.4.4 ENVIRONMENTAL REQUIREMENTS

Hard clams are most susceptible to changes in environmental parameters during their early stages of development. Tolerance limits to parameters such as temperature and salinity increase as the clams reach the adult stage (Arnold, pers. comm. 1992). This increased adult tolerance is a result of the clam's ability to isolate itself from ambient conditions by valve closure. This behavior, however, is only useful for surviving intermittent or short-term events such as hurricanes (Haven et al. 1977) or increased freshwater inflow from river discharges (Burrell 1977).

As with hard clam reproduction, most studies relating to environmental requirements of the hard clam were conducted using the northern quahog. Very little information exists on requirements of the southern quahog. In addition, very few studies were conducted specifically in Tampa Bay or with Tampa Bay clams. When possible, information was obtained from studies conducted in areas near Tampa Bay (e.g., the Indian River on the east coast of Florida or Alligator Harbor in northern Florida).

4.4.4.1 Temperature

Water temperature affects both reproduction and growth of the hard clam. Critical spawning temperatures vary according to geographic location. In the Indian River, both species of hard clam and their hybrids spawned when temperatures exceeded 18.5 °C and continued to spawn until water temperatures reached 28.3 °C (Hesselman et al. 1989).

As mentioned earlier, larval clams are less tolerant of temperature changes than adults. Davis and Calabrese (1964) observed that when water temperatures were around 10 °C, larval activity was minimal and growth was halted. They found that growth of the northern quahog straight-hinged veligers was positively related to temperature at 18.0 to 30.0 °C. Because of the physiological interactions of temperature and salinity, the optimum temperatures for northern quahog larval growth were reported in the following temperature/salinity combinations: 30 °C at 22.5 ppt and higher, 27.5 °C at 20 ppt, and 25 °C at 15 ppt (Davis and Calabrese 1964). Kennedy et al. (1974) observed that temperature tolerance increased with age; cleavage stages were the most sensitive to high temperatures and straight-hinged larvae were the least sensitive.

Ansell (1967) determined that the optimum temperature for adult northern quahog growth was 20°C and dropped off symmetrically at higher and lower temperatures, ceasing below 9°C and above 31°C. In northern Florida, Menzel (1961, 1962) demonstrated that growth of the southern quahog was minimal during the winter when temperatures approached 10 to 12°C. Southern quahog continues growth until reaching an upper temperature of 35°C (Mulholland 1984). Arnold et al. (1991) documented shell growth in hard clams from the Indian River, Florida, ceased at water temperatures of around

4.4.4.2 Salinity

Egg and larval stages of the northern quahog are more sensitive to salinity changes than adults (Stanley and DeWitt 1983). Davis (1958) conducted several studies on the effects of salinity changes on eggs and larvae of the northern quahog. He found the optimum salinity for egg development was about 26.5 to 27.5 ppt and they developed normally within a salinity range of 20 to 35 ppt. For normal larval development, the upper and lower salinity limits have been documented at 15 to 35 ppt (Loosanoff and Davis 1963); however, Castagna and Chanley (1973) found that metamorphosis of the northern quahog from veliger to juvenile stage was inhibited below 17.5 ppt.

Woodburn (1961, 1962) reported optimum salinity for the adult southern quahog was 35 to 36 ppt with 20 ppt as a minimum level. Turner (1953) and Chanley (1958) determined the optimum salinity for growth of the adult northern quahog was 24 to 28 ppt. Castagna and Chanley (1973) determined experimentally that the nonlethal minimum salinity for the adult northern quahog was 12.5 ppt (Castagna and Chanley 1973). In the Indian River, Florida, Arnold et al. (1991) found that both species of hard clams and their hybrids exhibited no clear relationship between the pattern of shell growth and ambient salinity which ranged from 20.1 to 25.3 ppt. In Tampa Bay, Taylor and Saloman (1970) suggested that the southern quahog prefers salinities greater than 24 ppt. Even if salinities exceed tolerance limits, both species of adult hard clams can tolerate extreme salinity changes for short periods of time by closing their shells. In a laboratory study, Pearse (1936) found that adult northern quahogs could survive in freshwater for approximately 114 hours.

The interaction between temperature and salinity can affect the tolerance limits of the hard clam to each of these parameters. For example, the range of temperatures tolerated by larval hard clams is reduced considerably when salinity is reduced (Eversole 1987).

4.4.4.3 Dissolved Oxygen

Normal larval development of the northern quahog will occur if dissolved oxygen (DO) concentrations are above 4.2 mg/L, whereas below this level, growth will gradually decrease, ceasing at 2.4 mg/L (Morrison 1971). Interestingly, Morrison (1971) found that embryos would develop normally at oxygen levels as low as 0.5 mg/L; however, 100% mortality occurred when DO levels dropped to 0.2 mg/L. Adult northern quahogs can maintain oxygen consumption in waters where DO equals or exceeds 5.0 mg/L. Below this level the hard clam begins undergoing oxygen stress, although activity can be maintained at DO concentrations less than 1.0 mg/L for up to 3 weeks (Savage 1976). In Tampa Bay, Godcharles and Jaap (1973) collected numerous southern quahogs in waters with DO values ranging from 4.0 to 7.8 mg/L.

4.4.4.5 pH

Optimum pH for northern quahog larvae ranges from 7.5 to 8.0, but normal growth will occur over the range of 6.75 to 8.50 (Calabrese and Davis 1966). Although clam larvae can survive at pH 6.25, a pH of 7.0 is required for normal development of the embryo. If pH levels drop below 7.0 northern quahog recruitment can be adversely affected (Calabrese 1972). For normal reproduction to occur in adult northern quahogs, Calabrese (1972) recommended that pH should not fall below 7.0 or above 9.0.

4.4.4.4 Suspended Solids

The eggs and larvae of hard clams can be adversely affected by heavy sediment loads. Davis (1960) found that at silt concentrations of 3.0 or 4.0 g/L clam eggs did not develop properly. Additionally, he found growth of straight hinged veligers was normal at a silt concentration of 0.75 g/L, but substantially retarded at 1.0 to 2.0 g/L. Bricelj et al. (1984) found that algal ingestion rate declined with increasing sediment loads. They determined that a significant reduction in growth and condition of juvenile northern quahogs occurred at silt concentrations of 0.044 g/L, but there was no effect up to 0.025 g/L. In Tampa Bay, high turbidities associated with silt-clay sediments in the extensively dredged portions of central and northern Boca Ciega Bay have limited the abundance and diversity of benthic mollusks, including the hard clam (Sykes and Hall 1970).

4.4.4.6 Substrate

Bottom substrate appears to be the primary factor responsible for the settling of hard clam larvae (Thorson 1955). Shelly substrates covered with a thin layer of detritus attract the largest sets of hard clams, followed by sandy sediments, with mud being the least colonized sediment (Carriker 1961; Keck et al. 1974). Pratt and Campbell (1956) found an inverse relationship between growth of the northern quahog and the fineness of the sediment. Newly set clams are negatively phototactic, and tend to accumulate in areas of current differentials such as quiet pockets within otherwise turbulent subtidal shell deposits (Wells 1957; Carriker 1959). In addition, laboratory experiments indicate that chemical exudates from clam larvae will enhance the attractiveness of a substrate to other plantigrade clams (Keck et al. 1974).

Adult hard clams are capable of living in a variety of sediment types. Rhoads and Panella (1970) determined that shell growth rate was significantly greater in the northern quahog living in sand than in mud. Hibbert (1976) found that growth rates of the northern quahog were similar on bottom substrates of 3% to 93% mud. Sims and Stokes (1967) reported no clear relationship between southern quahog density and sediment particle size in the Tampa Bay estuary; however, in Narragansett Bay, RI, Pratt (1953) established an inverse relationship between northern quahog clam density and particle size.

Several studies which were conducted in Florida's west coast estuaries including Tampa Bay have reported an association between the distribution of the southern quahog and stands of the seagrass, ***Thalassia testudinum*** (Schroeder 1924; Woodburn 1962; Sims and Stokes 1967; Taylor and Saloman 1968,1970; and Godcharles 1971). Wells (1957) suggested that seagrass reduced current speed, which favored increased settings of the hard clam. Kerswill (1941) suggested that seagrass provides attachment sites for the settling clam larvae. Seagrasses may also provide favorable clam habitat by entrapping and increasing deposition of silt and detritus, thereby binding and stabilizing the substrate (Ginsburg and Lowenstam 1958; Phillips 1960; Humm 1973). Although seagrasses may provide protection from predators and stabilize the sediment (Godcharles and Jaap 1973, Peterson 1982), they are apparently not essential for the well-being and survival of clams (Mulholland 1984).

4.4.4.7 Water **Depth**

Adult hard clams appear to prefer relatively shallow water, even though they also occur in the open ocean. Godcharles and Jaap (1973) reported numerous southern quahogs at moderate to shallow stations (less than 5 m) in Tampa Bay. Along the west coast of Florida the most productive beds were at 5.5 to 7.3 m. Southern quahogs were generally most abundant at depths of 4.7 to 9.2 m (Mulholland 1984).

4.4.4.8 Water Current

Water current is important for the growth and survival of hard clams; it provides food, acceptable water quality, removes biodeposits, and transports eggs and larvae (Belding 1931). Without adequate current velocity for proper flushing, areas can increase in silt and produce a undesirable soft sediment. In upland canals located in Tampa Bay, Sykes and Hall (1970) attributed the soft sediment caused by dredge and fill operations and inadequate flushing as the principal factor limiting the abundance and diversity of benthic mollusks. Carriker (1952) found clam larvae in water currents of 12 to 130 cm/sec and Kerswill (1949) reported adult clams grew better in water that had currents of 7.5 cm/sec compared to that of sluggish water.

Environmental requirements of hard clams are summarized in Table **4-6.**

4.5 GRASS SHRIMP *(Palaemonetes* spp.)

4.5.1 INTRODUCTION

Grass shrimp are ecologically important to the Tampa Bay estuary and are a vital link in the marine food web. Grass shrimp have been recognized as important prey for many bay species (Darnell 1958; McMichael and Peters 1989), in addition to playing a

Table 4-6. General and preferred ranges and upper and lower tolerance limits for environmental requirements of hard clam. Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				18.5-28.3 (S)	Hesselman et al. 1989
		10 (L)			Davis and Calabrese 1964
	Dependent on salinity (L)				Davis and Calabrese 1964
	20 (A)	9 (A)	31 (A)		Ansell 1967
		10-12 (A)			Menzel 1961, 1962
			35 southern (A)		Mulholland 1984
			30 (A)		Arnold et al. 1991
Salinity (ppt)	26.5-27.5 (E)			20-35 (E)	Davis 1958
		15 (L)	35 (L)		Loosanoff and Davis 1963
		17.5 (L,J)			Castagna and Chanley 1973
	35-36 southern (A)	20 (A)			Woodburn 1961, 1962
	24-28 northern (A)	12.5 (A)			Castagna and Chanley 1973
	>24 southern (A)				Taylor and Saloman 1970
Dissolved Oxygen (mg/l)	>4.2 northern (L)	2.4 (L)			Morrison 1971
		to 0.5 (L)			Morrison 1971
	75 northern (A)	to 1 (A)			Savage 1976
				4-7.8 southern (A)	Godcharles and Jaap 1973

Table 4-6. Continued

	Preferred	**Lower Limit**	**Upper Limit**	Range	Reference
PH	7.5-8 northern (L)			6.75-8.5 (L)	Calabrese and Davis 1966
	>7 northern (A)		9 (A)		Calabrase 1972
Current (cm/s)	12-130 (L)				Carriker 1952
Depth (m)				<5 northern (A)	Godcharles and Jaap 1973
				4.7-9.2 southern (L,J,A)	Mullholland 1984
Substrate	shelly, thin layer detritus (L)			sandy (L), mud bottoms	Carriker 1961; Keck et al. 1974
		silty (A)			Pratt and Campbell 1956
	seagrass southern (A)				Simms and Stokes 1967; Taylor and Saloman 1968, 1970; Godcharles 1971

major role in transferring energy between trophic levels within bay ecosystems (Welsh 1975).

4.5.1.1 Distribution

Grass shrimp, ***Palaemonetes*** spp., are very common in intertidal salt marshes and shallow subtidal seagrass beds of estuaries along the East and Gulf Coasts of the United States (Holthius 1952). The grass shrimp ranges from Nova Scotia to Texas (Williams 1965; Williams and Wigley 1977). Its broad distribution is likely related to its ability to adapt to wide fluctuations in temperature and salinity (Wood 1967; Morgan 1980; Alon and Stancyk 1982), and to low oxygen conditions in poorly flushed and high detritus habitats (Welsh 1975).

In the Gulf of Mexico, grass shrimp consist of five species; ***P. vulgaris, P. intermedius, P. pugio, P. paludosus, P. kadiakensis.*** These species are relatively similar in morphological characters and some may be found within the same habitats (Anderson, 1985). In Tampa Bay, the first four species listed above have been observed in varying abundances; ***P. pugio*** generally is found in areas of the bay with salinities ranging from 8 to 35 ppt, ***P. intermedius*** is more confined to the higher salinity waters (30 to 35 ppt) of the lower bay and open Gulf, ***P. paludosus*** is primarily found in the oligohaline areas of the upper rivers, and ***P. vulgaris*** appears to be relatively rare in Tampa Bay (Farrell, pers. com. 1992). ***Palaemonetes pugio*** and ***P. intermedius*** were the principal grass shrimp species present in the mid and lower portions of the estuary with ***P. vulgaris*** occurring in very low abundances (Fonseca, unpublished data 1992). Of the five species, P. ***pugio*** has probably received the most attention; however, no information is available for any of these species regarding their life history and ecology in Tampa Bay. Information will primarily be drawn from studies conducted in other estuaries.

4.5.2 LIFE HISTORY

4.5.2.1 Reproduction

The spawning season of the grass shrimp is variable depending on species and geographic location; however, grass shrimp generally spawn from February through October in Gulf of Mexico estuaries (Anderson 1985), with the exception of ***P. paludosus,*** which spawns year-round in southern Florida (Dobkin 1963). In Galveston Bay, Texas, the spawning season of ***P. pugio*** consists of two peaks; the first begins in early summer and the second in early fall (Wood 1967).

Sexually mature females molt into "breeding dress", which is characterized by the presence of extra setae on the pleopods, enlargement of the abdominal brood pouch, and development of periodic chromatophores (Antheunisse et al. 1971). They usually mate within a seven-hour period. Spawning usually takes place a few hours after mating and the fertilized eggs are carried on the females pleopods (Burkenroad 1947). The number of eggs a female produces varies (e.g., 300-500, Welsh 1975; Wood 1967), however a direct positive relationship between female length and number of eggs has been identified. Gravid ***P. pugio*** appear to migrate toward higher salinity waters to release their newly hatched larvae (McKenney 1979). Eggs hatch after a period of 12 to 60 days depending on the species and geographic location (Anderson 1985). Females have the potential to produce additional broods depending on the species and time of spawning. While carrying eggs, females may also have developing eggs (Broad and Hubschman 1963; Knowlton and Williams 1970; Beck and Cowell 1976).

4.5.2.2 Larval Development

Depending on the species and environmental conditions, there may be 3 to 11 morphologically distinct stages during larval development (Anderson 1983). Broad (1957) studied larval development of ***P. pugio*** and found that in water temperature of 25 and 28°C, the time and number of developmental stages varied depending on diet. For example, larvae fed Artemia metamorphosed in seven molts approximately two weeks after hatching whereas, larvae fed animal tissue and algae went through 13 molts over a period of two to four weeks. ***P. pugio*** larvae reared on Artemia at different salinities and temperatures went through different numbers of molts; at similar salinities, the number of molts were higher at lower temperatures (15°C) compared to those at higher temperatures (25°C) (Sastry 1980).

Morphologically, grass shrimp larvae lack long appendages and have to swim almost continuously with the head down and the dorsal surface oriented toward the direction of horizontal movement. Depending on environmental conditions, duration of ***P. pugio*** larval development may range from 11 days to several months (Floyd 1977).

Juvenile stages of the palaemonids, usually called postlarvae, are a transition stage between the pelagic larvae and benthic adults. Morphologically the postlarvae closely resemble adults (Anderson 1985). Kneib (1987) suggested that settlement of the planktonic larval stage and metamorphosis to the postlarval stage is triggered when the larvae enter a vegetated habitat. The juvenile stage lasts from 20 to 60 days (Piyatiratitivorakul 1988) and they develop into adults at about 15 to 18 mm TL (Alon and Stancyk 1982). Life span of adults is dependent on the season and environmental conditions of the specific estuary (Alon 1980). For example, the adult life span for ***P. pugio*** in a South Carolina estuary was estimated at less than one year by Sikora (1977) whereas Welsh (1975) estimated one year for the same species in Narragansett Bay, RI.

A summary of life history information for the five species of grass shrimp including spawning periods, fecundity, and larval development data is presented in Table **4-7.** Anderson (1985) described the life history of ***P. pugio*** from a southern coastal estuary. He reported that in the southern estuaries and coastal waters of the United States, overwintering ***P. pugio*** usually spawn from late February through March when water temperatures reach 15°C to 20°C. Juveniles from the February-March hatch then mature

Table 4-7. Spawning season, fecundity, lengths (mm) at hatching and at metamorphosis, and number of larval molts for ***Palaemonetes*** spp. in the Gulf of Mexico

Species	Spawning Season	Fecundity (Maximum observed)	Length (mm)[a]		No. of Larval Molts[a]
			at hatching	at meta-morphosis	
P. vulgaris	Apr-Oct[b]	No data	2.3	6.3	7-11
P. pugio	Mar-Oct[c]	486[d]	2.6	6.3	7-11
P. intermedius	May-Sep[b]	129[e]	3.5	7.0	6-8
P. kadiakensis	Feb-Oct[f]	160[g]	4.4	7.5	5-8
P. paludosus	Year round[h]	85[e]	3.8	4.5	3

[a] Data on length at hatching and metamorphosis and number of larval molts are from the following sources: ***P. vulgaris*** and ***P. pugio,*** Broad 1957; ***P. intermedius,*** Hubschman and Broad 1974; P. ***kadiakensis,*** Broad and Hubschman 1963; and ***P. paludosus,*** Dobkin 1963.

[b] Knowlton and Williams 1970

[c] Wood 1967

[d] Welsh 1975

[e] Beck and Cowell 1976

[f] White 1949

[g] Meehean 1936

[h] Dobkin 1963

Source: Anderson 1985

and spawn from late summer (July-August) to November, producing a second mode in the annual length distribution. Shrimp from the summer-fall hatch mature from October to February and reproduce during the following spring. Shrimp hatched in the spring have a faster growth rate than those hatched in summer and fall, however they do not live as long. Similar life history patterns of ***P. pugio*** probably occur in the Tampa Bay estuary as this species can be considered a resident of the bay.

4.5.3 ECOLOGICAL **ROLE**

4.5.3.1 **Food** Habits

Most palaemonid shrimp larvae are planktotrophic and their survival depends on the availability of zooplankton. The larvae cannot survive or undergo complete development if feeding is limited to plant materials or algae (Broad 1957a, b; Sandifer and Williams 1980). In contrast, juvenile and adult feeding behavior has been described as detritivorous (Welsh 1975), omnivorous (Morgan 1980) and carnivorous (Sikora 1977; Bell and Coull 1978; Kneib 1985). Juvenile and adult grass shrimp are also classified as predators and have been observed preying on meiofauna and small infaunal polychates, oligochaetes and nematodes (Sikora 1977; Bell and Coull 1978; Chambers 1981). Laboratory experiments conducted by Morgan (1980) suggest that predation on mysids is preferred to grazing. While living within aquatic macrophytes, shrimp feed on associated epiphytes which usually consist of filamentous and single-celled diatoms, green and blue-green algae, protozoans, rotifers and nematodes (Morgan 1980). Juvenile and adult grass shrimp can use epiphytes to satisfy all of their dietary demands.

4.5.3.2 Predation

Grass shrimp are preyed on by many types of estuarine animals, especially fish species. In Tampa Bay, grass shrimp have been found in the stomachs of economically valuable species such as spotted seatrout (Peebles 1992; McMichael and Peters 1989), snook (McMichael et al. 1989), and red drum (Peters and McMichael 1987). In other estuaries, forage fish such as ***Fundulus*** spp. (Harrington and Harrington 1972; Welsh 1975; Knieb and Stiven 1982) have been reported as major predators of grass shrimp. In addition, Kneib (1987) suggested that juvenile grass shrimp may be preyed upon by penaeid shrimp ***(Penaeus setiferus)*** in Georgia salt marshes.

4.5.4 ENVIRONMENTAL REQUIREMENTS

4.5.4.1 Temperature

Temperature and salinity properties are considered to be the major factors limiting the distribution of grass shrimp such as ***P. pugio*** (Wood 1967). Grass shrimp ***(P. pugio)*** have been collected at temperatures that range from 5 to 38°C, (Wood 1967; Christmas and Langley 1973), however, Wood (1967) suggested that optimum temperatures for survival of ***P. pugio*** in Galveston Bay, Texas were between 18 and 25°C. He also suggested that water temperatures of approximately 18 to 20°C were required for normal reproduction. Preferred water temperatures for ***P. vulgaris*** and ***P. paludosus*** range from 5 to 35°C and from 10 to 35°C respectively (Christmas and Langley 1973). A summary of temperature optima and tolerance limits for three species of grass shrimp including adult and larval forms was developed by Anderson (1985) and is presented in Table 4-8.

4.5.4.2 Salinity

Salinity is an important factor influencing the distribution of grass shrimp species. Salinity tolerance is variable and is primarily dependent on life stage and species. Several studies have determined that optimum salinity for ***P. pugio*** larvae is between 20 and 25 ppt (Floyd 1977; McKenney and Neff 1979; Knowlton and Kirby 1984). Studies have shown that extreme salinities of 16 and 46 ppt were lethal to 50% of ***P. pugio*** larvae (Kirby and Knowlton 1976); however, McKenney and Neff (1979) were able to rear larvae with at least 50% survival at salinities of 3 ppt. Larvae of ***P. vulgaris*** were able to tolerate salinities between 10 and 30 ppt, although 20 ppt was optimum (Sandifer 1973). ***Palaemonetes intermedius*** larvae were found to have similar salinity optima and tolerance limits as those reported for ***P. vulgaris*** (Broad and Hubschman 1962; Hubschman and Broad 1974). Larvae of the two freshwater species, ***P. paludosus*** and ***P. kadiakensis,*** could not survive at salinities above 5 ppt (Hubschman 1975; Strenth 1976). Salinity tolerance limits and optima were summarized by Anderson (1985) and are shown in Table 4-9.

In general, adult grass shrimp appear to have a greater tolerance to salinity changes than larvae. ***Palaemunetes pugio*** adults can tolerate salinities from 0 to 55 ppt, but are most common in salinities of 2 to 36 ppt (Wood 1967; Swingle 1971; Bowler and Seidenberg 1971; Christmas and Langley 1973; Kirby and Knowlton 1976; Morgan 1980). Wood (1967) suggested that salinities of 4 to 16 ppt were optimal for ***P. pugio*** survival. Adults of ***P. vulgaris*** have similar salinity requirements, however they are not as tolerant of low salinities (Holthuis 1952; Knowlton and Williams 1970; Bowler and Seidenberg 1971; Thorp and Hoss 1975; Nagabushanam 1961). Information on salinity requirements for ***P. intermedius*** is limited; however, Dobkin and Manning (1964) collected them from salinities that ranged from 5 to 39 ppt. Adults of the freshwater species, ***P. paludosus*** and ***P. kadiakensis,*** have been reported in salinities up to 30 (Swingle 1971) and 25 ppt, respectively (Maguire 1961; Strength 1976).

In mid to lower Tampa Bay, juvenile and adult ***P. intermedius, P. pugio,*** and ***P. vulgaris*** were collected in salinities that ranged from 18 to 45 ppt (Fonseca, pers. comm. 1992).

Table 4-8. Temperature tolerance range and optima (°C) reported for adult and larval *Palaemonetes.*

Species	Adults		Larvae	
	Limits	Optimum	Limits	Optimum
P. pauludosus	10-35[b]	18-33'	No data	No data
P. pugio	5-38[a,b]	18-25'	15-35[d,f]	20-30'
P. vulgaris	5-35[b]	No data	20-30"	20"

Sources:

[a] Christmas and Langley 1973
[b] Beck and Cowell 1976
[c] Wood 1967
[d] McKenney and Neff 1979
[e] Floyd 1977
[f] Sandifer 1973

Summarized by Anderson 1985

Table 4-9. Salinity tolerance limits and optima (ppt) reported by various investigators (see Anderson 1985) for adult and larval *Palaemonetes*

Form and Species	Adults		Larvae	
	Limits	Optimum	Limits	Optimum
Freshwater forms				
P. kadiakensis	0-25	0	0-10	0
P. paludosus	0-30	0	No data	0
Brackish-water forms				
P. intermedius	5-39	No data	10-30	20
P. pugio	1-55	4-16	3-31	25
P. vulgaris	1-51	No data	5-35	20

Source: Anderson 1985

4.5.4.3 Dissolved Oxygen

Limited information is available on dissolved oxygen (DO) requirements of grass shrimp. Rozas and Hackney (1984) found grass shrimp ***(P. pugio)*** were abundant in Cape Fear River (North Carolina) waters with DO levels of 2.8 to 4.4 ppm. In Louisiana, Barrett et al. (1978) found ***P. pugio*** and ***P. vulgaris*** in waters with DO ranging from 6 to 11 ppm. In laboratory studies, ***P. pugio*** had a greater tolerance to low DO concentrations (0.3 and 1.0 ppm) than ***P. vulgaris*** (Welsh 1975). Pomeroy and Wiegert (1981) noted that grass shrimp can avoid anoxic waters in confined areas by climbing out of the water for a few hours.

4.5.4.4 Turbidity

Grass shrimp are found in both high and low turbidity environments. Anderson (1985) suggested that grass shrimp may use turbid waters to avoid predation. Livingston et al. (1976) found that grass shrimp abundance was positively correlated with turbidity in Apalachicola Bay, Florida. However, high grass shrimp abundance was also reported, in clear water ponds in Louisiana (Weaver and Holloway 1974); although these ponds had dense aquatic vegetation which provided the grass shrimp protection from predators.

4.5.4.5 Depth

In general, grass shrimp are found in shallow coastal waters but can occur at depths up to about 16 meters (Williams 1965). The particular depth that grass shrimp inhabit is influenced by environmental factors such as temperature, salinity, and bottom type. Thorp (1976) noted that movement of grass shrimp between various depths coincided with the distribution of oyster shell substrate which is used by some species as a protective habitat.

4.5.4.6 Substrate

Many studies has documented the association of grass shrimp with aquatic vegetation such as seagrass and algae (Thorp 1976; Morgan 1980; Coen et al. 1981; Heck and Thoman 1981; Kneib 1987; Livingston et al. 1976). Grass shrimp use vegetated areas as nursery areas. These areas not only support an abundance and diversity of food for shrimp, but also provide a refuge from predators. It has been shown that grass shrimp are more prone to predation when displaced from protective macrophytes (Coen et al. 1981; Heck and Thoman 1981). In non-vegetated areas, oyster shells can be a preferred substrate of grass shrimp for similar reasons as mentioned for vegetated bottom (Thorp 1976).

Environmental requirements of ***P. pugio*** are shown in Table **4-10.**

Table 4-10. General and preferred ranges and upper and lower tolerance limits for environmental requirements of grass shrimp ***(P. pugio)*** Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, **J**=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature				5-38 (L,A)	Wood 1967, Christmas and Langley 1973
	18-25 (A)	**5 (A)**	**38 (A)**		Wood 1967
	18-20 **(S)**				Wood 1967
	20-30 (L)	**15 (L)**	**35 (L)**		McKenney and Neff 1979; Sandifer 1973
Salinity (ppt)	**20-25 (L)**	**3 (L)**	**31 (L)**		Floyd 1977; McKenney and Neff 1979; Knowlton and Kirby 1984
		16 (L) LD_{50}	**46 (L) LD_{50}**		Kirby and Knowlton 1976
		3 (L) LD_{50}			McKenney and Neff 1979
	4-16 (A)			1-55 (A)	Wood 1967; Swingle 1971; Bowler and Serdenberg 1971; Christmas and Langley 1973; Kirby and Knowlton 1976; Morgan 1980
Dissolved Oxygen **(mg/l)**				2.8-4.4 (A)	Rozas and Hacknev 1984
				6-11 (A)	Barrett **et** al. 1978
Depth **(m)**				to 16 **(J,A)**	Williams 1965
Substrate	aquatic vegetation seagrasses (J,A)				Thorp 1976; Morgan 1980; Coen et **al.** 1981; Heck and Thoman 1981; Knieb 1987; Livingston et al. 1976

5.0 BENTHIC COMMUNITIES

5.1 *DIOPATRA CUPREA*

5.1.1 INTRODUCTION

Benthic organisms play an important role in estuarine ecosystem by providing a linkage between primary producers and higher trophic levels for both planktonic and detritus-based food webs (Frithsen 1989; Holland et al. 1989) which ultimately produce many of the economically valuable fisheries found in the Tampa Bay estuary. The polychaete worm, *Diopatra cuprea* has been shown to be an important component of the Tampa Bay benthos by manufacturing tubes that provide important habitat for other benthic organisms (Bell and Coen 1982). However, as with many benthic invertebrates, only limited information is available on this species; studies primarily address species distribution, feeding habits, and environmental requirements as related to structural, habitat. Moreover, very little information currently exists regarding the environmental requirements of various benthic infauna found in Tampa Bay (Simon, pers. comm. 1992).

5.1.1.1 Distribution

Diopatra cuprea is generally found in intertidal and subtidal soft bottoms from the north side of Cape Cod, Massachusetts, to southern Florida (Mangum et al. 1968). In Tampa Bay, benthic studies have recorded the presence of ***D. cuprea*** at the following locations: Hillsborough Bay (Santos and Simon 1980), Apollo Beach located just south of Hillsborough Bay (Virnstein 1972), and Old Tampa Bay (Bloom et al. 1972; Simon and Dauer 1977; Bell and Coen 1982, Simon and Dauer 1972). ***Diopatra cuprea*** probably has a much wider distribution than identified from these studies, however relatively few baywide studies have been conducted in Tampa Bay, especially those which consider benthos.

5.1.2 LIFE HISTORY

Presently, no life history information on ***Diopatra cuprea*** is available (Bell, pers. comm. 1992), however Myers (1972) gives a description of their tube building process. The adult worm uses a mucous substance secreted from glands located between its segments to construct its tube. Once the mucous comes in contact with water it becomes hard. The tube consists of two different sections; an unreinforced tube which remains underground, and a reinforced tube which is above ground. As the worm moves through the sediment, it secretes mucous and the unreinforced tube develops. To build the reinforced tube, the worm, without leaving its underground tube, explores the sediment surface until it locates a piece of shell, pebble, or other object. Then it drags, pushes, or carries the object back to the tube and cements it into place near the opening of the unreinforced tube. The worm continues this process until the entire tube (unreinforced and reinforced) is approximately 2.5 cm long (range 1 to 6 cm). The tube then functions both as a refuge from predation and as a tool for catching food. If the reinforced, above-ground tube (tube cap) is destroyed by a predator or swept away by strong currents, the worm will rebuild it. Rebuilding time is temperature dependent; at 22°C, a new 1 cm tube cap can be built in approximately 24 hours.

5.1.3 ECOLOGICAL ROLE

5.1.3.1 Diet

Mangum et al. (1968) determined that ***D. cuprea*** has a varied diet consisting of both plant and animals, including; dinoflagellates, nematodes, annelids, copepods, and several species of algae. Interestingly, the authors concluded that the worm feeds largely, if not exclusively on living or recently deceased prey. A complete list of the stomach contents from ***D. cuprea*** collected in the Chesapeake Bay is presented in Table 5-1.

5.1.3.2 Predators

Because of its protective tube, ***D. cuprea*** can escape predation from many species; however, Myers (1972) noted that various fish and birds have been known to bite off the tube caps in attempts to feed on the worms. He also observed several sea stars ***(Asterias forbesi)*** attached to ***D. cuprea*** tube caps with their stomachs inverted. It was not clear whether the sea stars were actually feeding on the worms.

5.1.3.3 Habitat

As mentioned earlier, ***D. cuprea*** plays an important role in the ecosystem by providing habitat to other benthic organisms. Bell and Coen (1982) found primarily nematodes, copepods, and nauplii existing on ***D. cuprea*** tubes in Tampa Bay. Less abundant taxa included turbellarians, foraminiferins, amphipods, and juvenile and adult macrofauna such as the polychaete, ***Nereis succinea.*** Magnum et al. 1968 found similar fauna residing on tube caps in the Chesapeake Bay. Bell and Coen (1982) suggest that the production of tube caps by ***D. cuprea*** may be important in maintaining species abundance of tube inhabitants. In addition, species of copepods which use the tube caps for habitat and possibly breeding, are also found in the guts of some common benthic feeding fish such as ***Lagodon rhomboides*** (Stoner 1980) and ***Eucinostomas gula*** (Brook 1977), which lends support to the importance of worm tubes in the marine food web.

Table 5-1. Gut contents of Chesapeake Bay worms

- Protista
  - dinoflagellates
  - formaminiferan tests
  - unidentified flagellates
  - unidentified cyanophytes
- Nematoda
  - unidentified adults
  - capsules
- Annelida
  - ***Diopatra cuprea*** setae
  - clumps of capillary setae
  - uncinal setae
- Arthropoda
  - copepods
  - Unidentified appendages and spines
- Algae
  - Chlorophyta
    - ***Ulva*** sp.
    - unidentified multicelluar and unicellular forms
  - Rhodophyta
    - unidentified
  - Chrysophyta
    - unidentified
  - Angiospermae
    - ***Zostera marina***

Source: Mangum et al 1968

5.1.4 ENVIRONMENTAL REQUIREMENTS

Information concerning environmental requirements of ***D. cuprea*** is very limited. No studies have been conducted that address specific temperature, salinity, or dissolved oxygen optima or tolerance limits. However some abundance studies reported temperature and salinity at the time of collection. Some information was available on substrate preferences and water current requirements.

5.1.4.1 Temperature

In general, ***D. cuprea*** appears to tolerate a wide range of temperatures. Simon and Dauer (1977) observed ***D. cuprea*** in Tampa Bay at water temperatures that ranged from about 9 to 29°C. In the laboratory, Myers (1972) maintained ***D. cuprea*** at 0.5°C and noted that although the worms were surviving, activity such as tube maintenance ceased. He determined that temperatures must be above 1.8°C for tube maintenance to continue.

5.1.4.2 Salinity

Very little information is available to document salinity preference or limits of this species. ***Diopatra cuprea*** was observed in upper Tampa Bay waters where salinity ranged from 22.3 to 30.1 (Bloom et al. 1972; Bell and Coen 1982). However, distribution and salinity ranges in other portions of the estuary are unknown.

5.1.4.3 Substrate

Diopatra cuprea burrows into sediments ranging from sandy gravels to very fine-grained muds. Mangum et al. (1968) found about half of the population in the Cape Cod area occurred in bottoms with a modal grain size class of 250 to 499 μm, however a good correlation between abundance and grain size could not be established. In Tampa Bay, Bell and Coen (1982) observed worm tubes in sandy sediment that had a grain size of 21.

5.1.4.4 Depth

Diopatra cuprea has been documented to occur in both shallow and deep portions of the estuary. Santos and Simon (1980) found D. ***cuprea*** at depths of 4 to 5 m in Tampa Bay. Bell and Coen (1982) observed a high abundance of ***D. cuprea*** at a depth of 0.8 m in upper Tampa Bay. Virnstein (1972) found the worm tubes occurred at a depth of about 1 m at Apollo Beach in Tampa Bay.

5.1.4.5 Current

Current velocity is an important parameter for the survival of ***D. cuprea.*** It provides the worm with a food source, well-oxygenated water, and prevents increased sedimentation which could bury the worm tube. Myers (1972) observed that the tube cap of *D.* ***cuprea*** was oriented perpendicular to the current direction allowing a flow past the tube mouth for efficient feeding. Mangum et al. (1968) found a positive correlation between worm tube abundance and current velocity; large densities of *D.* ***cuprea*** were found in waters with current velocity ranging from 0.09 to 0.6 m/sec. In Tampa Bay, Santos and Simon (1980) collected ***D. cuprea*** at current velocities of about 0.5 knots.

Environmental requirements for ***D. cuprea*** are shown in Table 5-2.

Table 5-2. General and preferred ranges and upper and lower tolerance limits for environmental requirements of *Diopatra cuprea.* Letters in parentheses indicate life stage. S=spawning, E=egg, L=larval, J=juvenile, A=adult.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)				9-29 (A)	Simon and Dauer 1977
		1.8 (A)			Myers 1972
Salinity (ppt)				22.3-30.1 (A)	Bloom et al. 1972; Bell and Coen 1982
Current (m/s)				.09-.6 (A)	Mangum et al. 1968
Depth (m)				4-5 (A)	Santos and Simon 1980
				0.8-1 (A)	Bell and Coen 1982; Virnstein 1972
Substrate				sandy gravel to fine grained mud (A)	Mangum 1968, Bell and Coen 1982

6.0 MARINE MAMMALS

6.1 FLORIDA MANATEE *(Trichechus manatus latirostris)*

6.1.1 INTRODUCTION

The Florida Manatee (Trichechus manatus *latirostris)* is a distinct subspecies of the West Indian Manatee, one of four species which comprise the Order Sirenia (Reynolds and Odell 1991). In the United States, the manatee is considered an endangered species (USFWS 1991) and knowledge of critical habitats are essential to protecting this species.

The Florida manatee ranges as far north as Virginia and as far west as Mississippi or Louisiana during warm summer months. Its year round range is restricted to peninsular Florida, and possibly south Georgia (Reynolds and Odell 1991). The primary range of the Florida manatee is along the Atlantic coast of Florida from the St. Johns River in northeast Florida southward to the Miami area. Manatees are relatively abundant in the Indian River Lagoon area while fewer occur in the Florida Keys or Florida Bay. On the Gulf Coast, manatees are abundant from Everglades National Park northward to the Suwannee River (Reynolds and Odell 1991).

Manatees inhabit bays, estuaries, rivers and coastal areas where seagrasses and other vegetation are common. They rarely travel to deeper ocean waters and generally use these areas only as migratory routes between coastal regions (Hartman 1979).

6.1.2 LIFE HISTORY

Hartman (1979) investigated the life history of the manatee near Crystal River, FL. Manatees of both sexes reach maturity at ages of 6-10 years and approximately 2.5 to 2.7 meters in length. The manatee gestation period lasts approximately 12-13 months and usually only one calf is born; however, they may have twins. Calves are born primarily during spring and summer months and may stay with the females for one to two years. Female manatees produce calves every two to five years. Manatees are long-lived and may exceed fifty years of age (Hartman 1979).

Manatees are resident along the central part of the west coast of Florida in semi-isolated populations that are concentrated in rivers and estuaries that are of suitable depth and provide an adequate source of food and freshwater (Reynolds and Odell 1991). Manatee populations are concentrated in three regions; including the mouth of Suwannee River, spring-fed rivers in Citrus County, and the tidal tributaries in Tampa Bay (Reynolds and Odell 1991).

During the winter months, the Tampa Bay estuary contains approximately 8% (134 manatees) of the estimated minimum population of 1856 manatees in the United States (FDNR, unpublished data). The manatees in Tampa Bay are most abundant and aggregated in the winter when they gather in warm water discharges of power plants (Reynolds and Odell 1991).

Forty one manatee mortalities were verified in the Tampa Bay estuary during 1976-1985 (mean=4.1 /year)(Beeler and O'Shea 1988). From 1986-1990, 44 mortalities (mean=8.8/year) were observed (Reynolds et al. 1992; Table 6-1). A trend in decreasing percentage of calves has been noted in the Tampa Bay estuary (Table 6-2). Additional studies are needed to assess whether this perceived decline is due to lowered reproductive rates (Reynolds et al. 1992).

6.1.3 ECOLOGICAL ROLE

The Florida manatee is an opportunistic herbivore, feeding on a wide variety of plants, both aquatic and terrestrial, fibrous and nonfibrous and vascular and nonvascular (Hartman 1979). In saltwater, manatees feed primarily on several species of seagrasses, including turtle grass *(Thalassia testudinum),* manatee grass *(Syringodium filiforme),* and shoal grass *(Halodule wrightii).* They may also eat some species of algae, mangrove leaves and red mangrove seedlings. In fresh water, manatees feed on a variety of plants including *Hydrilla verticillata* , algae and water hyacinth (Reynolds and Odell 1991).

Some studies have suggested preferences for specific seagrasses. Hartman (1979) reported that manatees appeared to favor *Syringodium* where it grew in mixed stands of *Thalassia.* On the east central coast of Florida, manatee distribution data showed a positive correlation with *Syringodium* and *Halodule* and a negative correlation with *Caulerpa prolifera,* an attached macroalgae (Provancha and Hall 1991 b). These authors reported that this pattern suggested a selection for these seagrass dominated areas for feeding or potentially an avoidance of the areas containing large amounts of *Caulerpa.* Stomach content analyses of manatees in Brevard County indicated that *Caulerpa* was not an uncommon food item but it was found in relatively small amounts (Beck 1989).

Hartman (1979) mentioned that manatees may supplement their diet by feeding on macroalgae in areas where other vegetation was sparse. Manatees have been observed feeding on marine algae at the mouth of the Alafia River, and this algae may be an important food source for winter manatee populations. Dominant algae species in this area included *Gracilaria tikuahiae* (95%), *Ulva* spp. (4.8%) and *Chaetomorpha linum* (0.1%)(Lewis et al. 1984).

Some studies have documented seasonal movements of manatees along the west coast of Florida (Lefebvre and Frohlich 1986; Weigle et al. 1988). However, Reynolds et al. (1992) report that this may not be the general pattern and there was no strong evidence to indicate that manatees leave Tampa Bay during the summer. Radiotelemetry

Table 6-1. Manatee mortality by cause of death and year, for Tampa Bay area (Hillsborough, Manatee, Pinellas Counties), 1976-1990

	Year															
Cause of Death	**1976**	**1977**	**1978**	**1979**	**1980**	**1981**	**1982**	**1983**	**1984**	**1985**	**1986**	**1987**	**1988**	**1989**	**1990**	**Totd**
Watercraft collision			2		1		1		1	3	2		1	3	1	15
Flood gate/canal **lock**											2					2
Other human-related													1	1		2
Perinatal	1		2	2		1	1	1	2	2	2	4	4	3	5	30
Other natural						1		1	1	1		1	1	2	3	11
Undetermined	1	3	3	1	1	2	2	2		2	3				5	25
Total	2	3	7	3	2	4	4	4	4	8	9	5	7	9	14	85

Description of cause of death categories:

Watercraft collision: Manatees hit by boats, barges, or any other type of watercraft. Death may result from propeller wounds, impact, crushing, or a combination of the above.

Flood gate/canal lock: Manatees killed by crushing or asphyxiation in flood gates and canal locks.

Other human-related: Manatee deaths caused by vandalism, poaching, entrapment in pipes and culverts, complications due to entanglement in ropes, lines, and nets, or ingestion of fishing gear or debris.

Perinatal: Manatees up to 150 cm in total length which were not determined to have died due to any human-related causes.

Other natural: Manatee deaths resulting from infectious and noninfectious diseases, birth complications, natural accidents, and natural catastrophes (such as cold stress and red tide poisoning).

Undetermined: Manatee deaths in which the cause of death could not be determined. Rarely, deaths were reported and verified, but the carcass was not available for examination.

Source: Reynolds et al. 1992.

Table 6-2. Results of aerial surveys of manatees, by time periods, in Tampa Bay, Florida, November 1987 through December 1990

Season	Period	N	Mean	SE	Min	Max	% Calves
Warm	Nov 87	2	65.5	5.5	60	71	10.7
Cold	Dec 87 - Feb 88	6	49.8	13.7	28	88	11.0
Warm	Mar 88 - Nov 88	16	50.3	3.9	26	74	8.0
Cold	Dec 88 - Feb 89	6	58.7	8.5	31	81	8.2
Warm	Mar 89 - Nov 89	8	29.8	3.7	17	43	9.2
Cold	Dec 89 - Feb 90	3	76.3	6.4	69	89	7.4
Warm	Mar 90 - Nov 90	8	48.1	7.7	15	81	7.3
Cold	Dec 90	2	70.5	30.5	40	101	6.4
Total		51	50.6	2.9	1.5	101	8.4

Source: Reynolds et al. 1992

studies began in 1991 and will yield additional information on manatee movement patterns in Tampa Bay and into adjacent regions (Reynolds et al. 1992).

6.1.3.1 Predators

The manatee has no known predators. It appears that cold weather, shoreline development, injuries from boat collisions, and possibly pollution are among the factors limiting the survival of manatees in Florida (Schomer and Johnson 1990).

6.1.4 CONTAMINANTS

Some information was available to document contaminant concentrations in manatee tissues. O'Shea et al. (1984) reported that copper buildup may result from feeding on vegetation in areas where copper based herbicides have been used to control aquatic vegetation. Copper residue concentrations in livers of manatees from high copper herbicide use areas near Crystal River (Citrus, Dixie and Levy Co.) were greater than from medium (Brevard and Dade, Co.) or low copper use areas (Broward, Collier, Hillsborough, Lee, Martin, Monroe and Palm Beach) (O'Shea et al. 1984). These authors reported that since high copper concentrations have been linked to toxic effects in domestic mammals, it may also be harmful to manatees and the use of copper-based aquatic herbicides should be carefully managed in areas used intensively by manatees.

Analysis of blubber samples from 26 manatees indicated that these animals are relatively free from contamination by organochlorines. Five of 26 samples contained detectable residue concentrations of the metabolites of DDT (mean=0. 19 +- 0.10 ppm DDTR; range=0.14-0.28 ppm). Dieldrin was detected in 4 of 26 samples (mean=0.26 ± 0.10 ppm, range=0.12-0.36), and PCB's were detected in 13 of 26 individuals (mean=1.4 +- 1.1 ppm, range = 0.50-4.6), primarily from urbanized northeast Florida (lower St. Johns River and Brevard County)(O'Shea et al. 1984). Low levels of these contaminants are most likely a result of the unique food habits and the well known findings of decreased exposure to these substances lower (e.g. herbivores) in the food chain.

6.1.5 ENVIRONMENTAL REQUIREMENTS

Environmental requirements of the Florida manatee have been fairly well documented in the Tampa Bay estuary. Three critical manatee requirements include: warm water in the winter months, fresh water, and abundant seagrasses for food (Reynolds et al. 1992). Specific requirements and limiting factors will be discussed in the following sections.

6.1.5.1 Salinity

Access to fresh water is an important aspect of manatee distribution in Florida (O'Shea and Kochman 1990). Manatees are frequently seen in areas where rivers, drainage canals, sewer outfalls, or other sources of fresh water predominate. Manatees move freely between fresh and salt water, although they prefer waters with salinities less than 25 ppt (Hartman 1979). This author reported that manatees observed in high salinity, open ocean or Gulf waters appeared to be migrating.

Throughout the range of this species, strictly marine habitat (salinities > 30 ppt) is not frequently occupied (Lefebvre et al. 1989; cited from O'Shea and Kochman 1990). It is likely that for reasons of metabolic economy, manatees prefer to occupy habitats where osmotic stress is minimal or where periodic freshwater drinking is feasible (O'Shea and Kochman 19903, they can; however, persist for periods in salt water. Manatees can live indefinitely in fresh water, however, the maximum duration in salt water is not known. They require fresh water for drinking (Reynolds and Odell 1991; Reynolds et al. 1992). Therefore, it appears that while the Florida manatee can tolerate a wide range of salinities, they will need access to areas of fresh water.

6.1.5.2 Temperature

Manatee distribution is affected by seasonal temperature changes. In response to water temperature declines, the population shifts southward to warmer waters. During winter months manatees congregate at natural warm water springs on the Gulf coast and in regions of warm water discharges from power and other industrial plants (Reynolds and Odell 1991). Reynolds et al. (1992) used aerial surveys to document seasonal distribution patterns of manatees in Tampa Bay. Manatees aggregated at warm water discharges from December-February (and to a lesser extent in adjacent months) and then dispersed widely throughout the bay. During warm months, manatees are found in many areas around Tampa Bay and the tributary rivers. Lower manatee counts occurred during summer months, although these may have reflected seasonal differences in visibility rather than emigrations of manatees from the survey area. Highest survey counts during summer months occurred in the Old Tampa Bay, Big Bend, Terra Ceia Bay, north St. Petersburg and Manatee River areas. These areas comprised 77% of the total manatees in the Tampa Bay estuary (Reynolds et al. 1992).

It appears that manatee thermal neutrality reaches a lower limit in a range of 20-24oC, and when ambient water temperatures remain consistently below 20°C, manatees move to sources of warm water (Hartman 1979; Bengston 1981; Irvine 1983; cited from O'Shea and Kochman 1990). Hartman (1979) observed manatees feeding casually in lower Tampa Bay in water as cold as 19°C and reported that they endure water temperatures at least as cold as 13.5°C. Thermal stress from prolonged exposure to cold water temperatures can result in manatee mortalities (O'Shea et al 1985). Five manatees were killed in Charlotte Harbor, FL when water temperatures dropped from 20° to 8°C during a winter freeze in 1940 (Cahn 1940). Over 50 manatee deaths occurred during the harsh winter freezes in December 1989 (FDNR, unpublished data).

Manatees may find it more energetically advantageous to remain in warm water areas without feeding for up to a week rather than venturing into colder waters for food (Bengtson 1981). Warm water refuges in the Tampa Bay estuary include: Port Sutton power station, Big Bend power station and the Bartow power station (Reynolds et al. 1992). Manatee counts in zones containing those three power plants included 84% of manatees sighted during cold weather surveys.

Irvine and Campbell (1978) report that the impact of artificial warm water sources on manatee survival and winter distribution is unknown. The suitability of artificially warmed habitats has been questioned. Increased availability of these warm water sources may have altered historic winter distribution patterns of manatees by diverting animals from southward winter migrations (Campbell and Powell 1976).

6.1.5.2 Water Depth

Hartman (1979) reported that the range of the manatee on the east and west coasts of Florida is delimited by shallow water. In the Crystal River area, manatees carried out most of their activities in water two to three meters deep and normally travelled via waterways that were at least two meters deep. Flats and shallow areas that were less than one meter were avoided unless immediately adjacent to deeper water. In Tampa Bay, manatees were observed to favor water 1.5-2.0 m deep (Hartman 1979).

6.1.5.3 Structural Habitat

Manatees inhabit bays, estuaries, rivers and coastal areas where seagrasses and other vegetation are common (Reynolds and Odell 1991). Manatees feed primarily on submerged aquatic vegetation; therefore, areas containing adequate amounts of these resources should be considered critical habitats.

Rather than any single environmental requirement being critical to manatee survival, the interaction of water quality and structural habitat parameters probably best describe critical habitat of the manatee. In addition to seagrass and other aquatic vegetation which fulfill feeding requirements, manatees also require a source of fresh water for drinking and a warm water refuge for winter months (Reynolds et al. 1992). These authors reported areas within Tampa Bay which provided critical habitat, including: 1) warm water outfalls at Bartow, Big Bend and Port Sutton power plants, and the Cargill Fertilizer Company (previously Gardinier Phosphate Co.), 2) Coffeepot Bayou, 3) Hillsborough River, 4) Portions of the Little Manatee and Manatee Rivers, 5) Braden River, 6) Terra Ceia Bay and 7) Anna Maria Sound. More recent data suggests that manatee use of the Cargill Fertilizer Company outfall waters has greatly diminished (FDNR, unpublished data).

Environmental requirements of manatees are summarized in Table 6-3.

Table 6-3. General and preferred ranges and upper and lower tolerance limits for environmental requirements of juvenile and adult manatee. Letters in parentheses indicate life stage.

	Preferred	Lower Limit	Upper Limit	Range	Reference
Temperature (°C)	>20°C				O'Shea and Kochman 1990
		13.5oC			Hartman 1979
Salinity (ppt)	<25	0	33		Hartman 1979
				Freshwater to marine	Hartman 1979
Depth (m)	1.5-2 m	.7			Hartman 1979; FDNR, unpublished data
Substrate	Seagrasses or other aquatic vegetation			Various	Reynolds and Odell 1991; Hartman 1979; FDNR, unpublished data

7.0 REFERENCES

Aaron, R.L. and Warren J. Wisby. 1964. Effects of light and moon phase on the behavior of pink shrimp. Proc. Gulf Carib. Fish. Inst. 16th Annu. Sess.: 121-130.

Abbott, R.T. 1954. American Seashells. New York: D. Van Nostrand Co., Inc. 541p.

Abbott, R.T. 1974. American Seashells. 2nd ed. New York: Van Nostrand Reinhold Co., Inc.

Abbott, R.T. 1974. American seashells. The marine Mollusca of the Atlantic and Pacific coasts of North America. 2nd ed. New York: Van Nostrand Reinhold Co.

Abraham, B.J. 1985. Species Profiles: Life Histories and Environmental Requirements of Coastal Fishes and Invertebrates (Mid-Atlantic) - Mummichog and Striped Killifish. U.S. Fish and Wildl. Serv. Rep. 82(11.40).

Adkins, G. 1972. Notes on the occurrence and distribution of the rhizocephalan parasite *lLoxothylacus texanus* Boschma) of blue crabs *Callinectes sapidus* Rathbun) in Louisiana estuaries. *La. Wild/. Fish. Comm. Tech. Bull.* 2, 13 p.

Adkins, G. 1982. The blue crab fishery of Louisiana. *In:* H.M. Perry and W.A. Van Engel (editors), Proceedings Blue Crab Colloquium, October 18-19, 1979, p. 15-16.

Ahmed, M. 1975. Speciation of living oysters. *Adv. Mar. Biol.* 13:357-397.

Alexander, S.K. 1986. Diet of the blue crab, *Callinectes sapidus* Rathbun, from nearshore habitats of Galveston Island, Texas. *Texas J Sci.* 38:85-89.

Allen, D.M. and A. Inglis. 1958. A pushnet for quantitative sampling of shrimp in shallow estuaries. *Limnol. Oceanogr.* 3(2):239-241.

Alon, N.C. and S.E. Stancyk. 1982. Variation in life-history patterns of the grass shrimp *Palaemonetes pugio* in two South Carolina estuarine systems. *Mar. Biol.* 68:265-276.

American Fisheries Society (AFS). 1991. Common and Scientific Names of Fishes from the United States and Canada. 5th ed. AFS Special Publ. 20. Bethesda, MD.

Anderson, G. 1985. Species profiles: Life histories and environmental requirements of coastal fishes and invertebrates: grass shrimp (Gulf of Mexico). Biol. Rep. 82:11-35, TR-EL-82-4.

Andrews, J.D. 1979. Pelecypoda: Ostreidae. In: Reproduction of marine invertebrates. Vol. V. Molluscs: pelecypods and lesser classes, 291-341. A.C. Giese and J.S. Pearse, eds. New York: Academic Press.

Andrews, J.D. 1982. Transport of bivalve larvae in the James River, Virginia. Virginia Institute of Marine Science Special Report **111**:1-75.

Ansell, A.D. 1967. Egg production of *Mercenaria mercenaria* . *Limnol. Oceanogr.* 12:172-176.

Antheunisse, L.J., J.J. Lammens, and N.P. Van den Hoven. 1972. Diurnal activities and tidal migration of the brackish water prawn *Palaemonetes varians* (Leach) (Decapoda, Caridea). *Crustaceana* 21:203-217.

Archambault, J. and R.J. Feller. 1991. Diel variations in gut fullness of juvenile spot, *Leiostomus xanthurus* (Pisces). *Estuaries* 14:94-101.

Arnold, C.R., J.L. Lasswell, W.H. Bailey, T.D. Williams, and W.A. Fable, Jr. 1976. Methods and techniques for spawning and rearing spotted seatrout in the laboratory. Proc. Annu. Conf. Southeast Assoc. Game Fish Comm. 30:167-178.

Arnold, E.L. and J.R. Thompson. 1958. Offshore spawning of the striped mullet, *Mugil cephalus,* in the Gulf of Mexico. *Copeia* 130-132.

Arnold, W. 1992. Personal communication from W. Arnold, Florida Department of Natural Resources, Florida Marine Research Institute, to R. Hochberg, January 1992.

Arnold W.S. 1984. The effects of prey size, predator size, and sediment composition on the rate of predation of the blue crab, *Callinectes sapidus* Rathbun, on the hard clam, *Mercenaria mercenaria* (Linne). *J. Exp. Mar. Biol. Ecol.* 80:207-219.

Arnold, W.S., D.C. Marelli, T.M. Bert, D.S. Jones, and I.R. Quitmyer. 1991. Habitat specific growth of hard clams *Mercenaria mercenaria* (L.) from the Indian River, Florida. *J Exp. Mar. Biol. Ecol.* 147:245-265.

Bahr, L.M., Jr. 1976. Energetic aspects of the intertidal oyster reef community at Sapelo Island, Georgia. *Ecology* 57(1):121-131.

Bahr, L.M. and W.P. Lanier. 1981. The ecology **of** interidal oyster reefs of the south Atlantic coast: a community profile. U.S. Fish Wildl. Serv. Biol. Serv. Program FWS/OBS-8**1/**15. 105 **p.**

Baird, D. and R.E. Ulanowicz. 1989. The seasonal dynamics of the Chesapeake Bay ecosystem. *Ecol. Monog.* 59:329-364.

Baker-Dittus, A.M. 1978. Foraging patterns of three sympatric killifish. *Copeia* 1978(3):383-389.

Banks, M.A., G.J. Holt, and J.M. Wakeman. 1991. Age-linked Changes in Salinity Tolerance of Larval Spotted Seatrout *(Cynoscion nebulosus,* Cuvier). *Journal of Fish Biology* 39:505-514.

Barrett, B.B., J.L. Merrell, T.P. Morrison, M.C. Gillespie, E.J. Ralph, and J.F. Burdon. 1978. A study of Louisiana's major estuaries and offshore waters. *La. Dep. Wild/, Fish. Tech. Bull .* 27:1-197.

Bass, R.J. and J.W. Avault, Jr. 1975. Food habits, length-weight relationship, condition factor, and growth of juvenile red drum, *Sciaenops ocellata ,* in Louisiana. *Trans. Am. Fish; Soc.* 104:35-45.

Bateman, H.A. 1965. Clapper rail *(Rallus longirostris)* studies on Grand Terre Island, Jefferson Paris, Louisiana. Master's thesis. Louisiana State University, Baton Rouge, LA. 145 p.

Baugman, J.L. 1945. Some serranid fishes of Texas: the Centropomidae, Moronidae, and Epinephelidae. *Am. Mid. Nat.* 30(3):769-773.

Beaumariage, D.S. 1969. Returns from the 1965 Schlitz tagging program including a cumulative analysis of previous results. *Fla. Bd. Conserv. Mar. Res. Lab. Tech. Ser.* No. 59, 38 p.

Beaumariage, D.S. and A.C. Wittich. 1966. Returns from the 1964 Schlitz tagging program. *Fl. Bd. Conserv. Mar. Res. lab. Tech. Ser .* 47:50 pp.

Beck, C. 1989. Pers. Comm., United States Fish and Wildlife Service, Denver Wildlife Research Center, Gainesville, Florida.

Beck, J.T. and B.C. Cowell. 1976. Life history and ecology of the freshwater caridean shrimp, *Palaemonetes paludosus* (Gibbes). *Am. Midl. Nat .* 96:52-65.

Beebe, W. 1927. A Tarpon nursery in Haiti. *Bull. NY Zool. Soc .* 30:141-145.

Beeler, I.E. and T.J. O'Shea. 1988. Distribution and mortality of the West Indian manatee *(Trichechus manatus)* in the United States: A compilation and review of recent information. Volume Two: The Gulf of Mexico coast. pp. 329-617. *In: Natl. Ecol. Res. Ctr. Rept.* No. 88-09. NTIS PB88-207980.

Belding, D.C. 1912 (reissued 1964). A report upon the quahaug and oyster fisheries of Massachusetts, including the life history, growth, and cultivation of the quahaug *(Venus mercenaria) ,* and observations on the set of oyster spat in Wellfleet Bay. Mass. Div. Mar. Fish., Contrib. No. 12. 134p.

Belding, D.L. 1911. The life history and growth of the quahaug *(Venus mercenaria).* Rep. Mass. Comm. Fish Game 1910:18-128.

Belding, D.L. 1931. The quahaug fishery of Massachusetts. The Commonwealth of Massachusetts, Marine Fishery Series 2: 1-41.

Bell, S. 1992. Personal communication from S. Bell, University of South Florida, Tampa, Florida, to R. Hochberg, November 1992.

Bell, S.S. and B.C. Coull. 1978. Field evidence that shrimp predation regulates meiofauna. *Oecologia* 35:141-148.

Bell, S.S. and L.D. Coen. 1982. Investigations on epibenthic meiofauna **I.** Abundances on repopulation of the tube-caps of *Diopatea cuprea* (Polychaeta-Onophidae) in a subtropical system.

Bender, M.E., W.J. Hargis, Jr., R.J. Huggett, and M.H. Roberts, Jr. 1988. Effects of polynuclear aromatic hydrocarbons on fishes and shellfishes: An overview of research in Virginia. *Mar. Environ. Res.* 24:237-241.

Bengston, J.L. 1981. Ecology of manatees *(Trichechusmanatus)* in the St. Johns River, Florida. Ph.D. Thesis, University of Minnesota, Minneapolis. 126 p.

Bent, A.C. 1926. Life histories of North American marsh birds. U.S. Natl. Mus. Bull. (Smithsonian Inst.) 135. 490p.

Berrien, P.L., M.P. Fahay, A.W. Kendall, Jr., and W.G. Smith. 1978. Ichthyoplankton from the RV *Dolphin* survey of the Continental Shelf waters between Martha's Vineyard, Massachusetts and Cape Lookout, North Carolina, 1965-66. *Natl. Mar. fish. Serv. Sandy Hook Lab. Tech. Ser. Rep. No.* 15. 152 pp.

Berrigan, M. 1992. Personal communication from M. Berrigan, Florida Department of Natural Resources, Tallahassee, Florida, to R. Hochberg, June 1992.

Berrigan, M.E. 1990. Biological and economical assessment of an oyster resource development project in Apalachicola Bay, Florida. *J Shellfish Res.* 9:149-158.

Berrigan, M., T. Candies, J. Cirino, R. Dugas, C. Dyer, J. Gray, T. Herrington, W. Keithly, R. Leard, J.R. Nelson, M. Van Hoose. 1991. The oyster fishery of the Gulf of Mexico, United States: A regional management plan. Gulf States Marine Fisheries Commission, No. 24.

Bielsa, L.M., W.H. Murdich, R.F. Labisky. 1983. Species profiles: Life history and environmental requirements of coastal fishes and invertebrates pink shrimp (south Florida). U.S. Fish and Wildl. Sew. FWS/OBS-82/11-17; TR EL-82-4.

Bigelow, H.B. and W.C. Schroeder. 1953. Fishes of the Gulf of Maine. U.S. Fish and Wildl. Serv. *Fish. Bull.* 53:1-577.

Birdsong, R.S. 1981. A review of the gobiid genus *Microgobius Poey. Bull. Mar. Sci.* 31:267-306.

Blooms, S.A., J.L. Simon, and V.D. Hunter. 1972. Animal-sediment relations and community analyses of a Florida estuary. *Mar. Biol.* 13:43-56.

Bookhout, C.G. and J.D. Costlow, Jr. 1975. Effects of mirex on the larval development of blue crab. *Water, Air, Soil Pollut.* 4:113-126.

Bookhout, C.G., J.D. Costlow, Jr., and R. Monroe. 1976. Effects of methoxychlor on larvae development of mud-crab and blue-crab. *Water, Air, Soil Pollut* . 5:349-365.

Boothby, R.N. and J.W. Avault, Jr. 1971. Food habits, length-weight relationship and condition factor of the red drum *(Sciaenops ocellata)* in southeastern Louisiana. *Trans. Am. Fish. Soc.* 100(2):290-295.

Botello, A.V., S.A. Castro, and L. Juarez M. 1981. Efecto de **los** hidrocarburos fosiles en las comunidades del genero *Penaeus* del Banco de Campeche, Mexico. *Proc. Gulf Caribb. Fish. Inst.* 33:69-80.

Bowler, M.W., and A.J. Seidenberg. 1971. Salinity tolerance of the prawns, *Palaemonetes vulgaris* and *P. pugio* and its relationship to the distribution of these species in nature. *Va. J. Sci.* 22:94.

Bozeman, E.L., Jr. and J.M. Dean. 1980. The abundance of estuarine larval and juvenile fish in a South Carolina intertidal creek. *Estuaries* 3:89-97.

Breder, C.M., Jr. 1933. Young tarpon on Andros Island. Bull. *NY Zool. Soc . 36:65-67.*

Breder, C.M., Jr. 1940. The spawning of *Mugil cephalus* on the Florida west coast. *Copeia* 138-139.

Breder, C.M., Jr. 1944. Materials for the study of the life history of *Tarpon atlanticus. Zoologica* 29:217-252.

Bricelj, V.M. and **R E.** Malouf. 1980. Aspects of reproduction of hard clams *(Mercenaria mercenaria*) in Great South Bay, New York. *Proc. Natl. Shellfish. Assoc.* 70:216-229.

Bricelj, V.M., R.E. Malouf, and C. DeQuillfeldt. 1984. Growth of juvenile *Mercenaria mercenaria* and the effect of resuspended bottom sediments. *Mar. Biol.* 84:167-173.

Bridges, D.W. 1971. The critical thermal maximum of juvenile spot, *Leiostomus xanthurus.* Lacepede. Water Res. Research Inst. Rep. No. 43. Univ. of North Carolina, p. 46.

Briggs, J.C. 1958. A list of Florida fishes and their distribution. *Bull. F1l St. Mus.* 2:225-318.

Broad, A.C. 1957. Larval development of *Palaemonetes pugio* Holthuis. *Biol. Bull.* (Woods Hole) 112:162-170.

Broad, A.C. 1957a. Larval development of *Palaemonetes pugio* Holthuis. Biol. *Bull. Mar. Biol. Lab.* , Woods Hole 112:144-161.

Broad, A.C. 1957b.- The relationship between diet and larval development of *Palaernonetes. Biol. Bull. Mar. Biol. Lab* ., Woods Hole 112:162-170.

Broad, A.C. and J.H. Hubschman. 1962. A comparison of larvae and larval development of species of Eastern U.S. *Palaemonetes* with special reference to the development of *Palaemonetes intermedius* Holthuis. *Am. Zool.* 2:394-395.

Broad, A.C. and J.H. Hubschman. 1963. The larval development of *Palaemonetes kadiakensis* M.J. Rathbun in the Laboratory. *Trans. Am. Microsc. Soc.* 82:185-197.

Broadhead, G.C. 1953. Investigations of the black mullet, *Mugil cephalus L,* in northwest Florida. *Fla. Board Conserv. Tech. Ser.* No. 7. 33 p.

Broadhead, G.C. and H.P. Mefford. 1956. The migration and exploitation of the black mullet, *Mugil cephalus* L. in Florida, as determined from tagging during 1949-1953. *Fla. Bd. Conserv. Tech. Ser.* No. 18. 31 p.

Brook, I.M. 1977. Trophic relationships in a seagrass community *(Thalassia testudinum),* in Card Sound, Florida. Fish diets in relation to macrobenthic and cryptic faunal abundance. *Trans. Am. Fish. Soc* . 106:219-229.

Brown, N.J. 1981. Reproductive biology and recreational fishery for spotted seatrout, *Cynoscion nebulosus,* in the Chesapeake Bay area. M.A. thesis, College of William and Mary, Williamsburg, VA. 119 p.

Brown, S. 1992. Personal communication from S. Brown, Florida Department of Natural Resources, Florida Marine Research Institute, St. Petersburg, FL to R. Hochberg, January 1992.

Bruger, G. 1992. Personal communication from G. Bruger, Florida Department of Natural Resources, Florida Marine Research Institute, St. Petersburg, FL to K. Killam, February 1992.

Buckley, J. 1984. Habitat Suitability Index Models: Larval and Juvenile Red Drum. USFWS FWS/OBS-82/10.74.

Bureau of Economic and Business Research. 1988. Population studies, January 1988. Univ. of Fla. Gainesville.

Bureau of Economic and Business Research (BEBR). 1988. Population studies Jan 1988. Univ. of FL, Gainesville.

Burkenroad, M.D. 1947. Reproduction activities of decapod Crustacea. *Am. Nat.* 81 :392-398.

Burkenroad, M.D. 1949. Occurrence and life histories of commercial shrimp. *Science* 110:688-689.

Burrell, V.G., Jr. 1977. Mortalities of oysters and hard clams associated with heavy runoff in the Santee River system, South Carolina, in the spring of 1975. *Proc. Natl. Shellfish. Assoc.* 67:35-43.

Burton, D.T., L.B. Richardson, and G.J. Moore. 1980. Effect of oxygen reduction rate and constant low dissolved oxygen concentrations on two estuarine fish. *Trans. Am. Fish. Soc.* 109:552-557.

Butler, P.A. 1949. An investigation of oyster producing areas in Louisiana and Mississippi damaged by flood waters in 1945. *U.S. Fish Wild/. Serv. Spec. Sci. Rep. Fish.* No. 8:1-27.

Butler, P.A. 1953. The southern oyster drill. Proc. *Natl. Shellfish. Assoc.* 44:67-75.

Butler, P.A. 1954. Summary of our knowledge of the oyster in the Gulf of Mexico. U.S. Fish Wildl. *Serv. Fish. Bull.* 55(89):479-489.

Butler, P.A. 1969. The significance of DDT residues in estuarine fauna. *In:* M.W. Miller and G.B. Berg (editors), Chemical Fallout, p. 205-220 . Charles C. Thomas Publishers, Springfield, IL.

Butler, P.A., R. Childress, and A.J. Wilson, Jr. 1970. The association of DDT residues with losses in marine productivity. FAO Technical Conference on Marine Pollution and its Effects on Living Resources and Fishing, 13 p.

Byrne, C.J. and J.A. Calder. 1977. Effect of the water-soluable fractions of crude, refined and waste oils on the embryonic and larval stages of the quahog clam, *Mercenaria* sp. *Mar. Biol.* 40:225-231.

Cahn, A.R. 1940. Manatees and the Florida freeze. *J. Mamm.* 21 :222-223.

Cake, E.W., Jr. 1983. Habitat suitability index models: Gulf of Mexico American oyster. U.S. Dept. Int. Fish Wildl. Serv. FWS /OBS-82/10.57. 37pp.

Calabrese, A. 1972. How some pollutants affect embryos and larvae of American oyster and hard-shell clam. *U.S. Natl. Mar. Fish. Serv. Mar. Fish. Rev.* 34(11-12):66-77.

Calabrese, A and H.C. Davis. 1966. The pH tolerance of embryos and larvae of *Mercenaria mercenaria* and *Crassostrea virginica. Biol. Bull.* 131:427-436.

Calabrese, A. and H.C. Davis. 1969. Spawning of the American oyster, *Crassostrea virginica* , at extreme pH levels. *Veliger* 11:235.

Calabrese, A. and H.C. Davis. 1970. Tolerances and requirements of bivalve molluscs. *Helgol. Wiss. Meeresunters .* 20:553-564.

Calabrese, A., E. Gould, and F.P. Thurberg. 1982. Effects of toxic metals in marine animals of the New York Bight: Some laboratory observations. *In: Ecological Stress and the New York Bight: Science and Management, 28 1-29 7.* G.F. Mayer, ed. Estuarine Res. Fed. Columbia, North Carolina.

Calabrese, A., J.R. MacInnes, D.A. Nelson, and J.E. Miller. 1977b. Survival and growth of bivalve larvae under heavy-metal stress. *Mar.* Biol. 41:179-184.

Calabrese, A., F.P. Thurberg, and E. Gould. 1977a. Effects of cadmium, mercury, and silver on marine animals. *Mar. Fish. Rev.* 39:5-11.

Call, B.C. 1985. Long-term variability of estuarine meiobenthos: a four-year study. Mar. Ecol Prog. Ser. 24:205-218.

Campbell, H.W. and J.A. Powell. 1976. Endangered species: The manatee. *Flor. Nat.* 49:15-20.

Cardeilnac, P.T., C.F. Simpson, F.H. White, N.P. Thompson, and W.E. Carr. 1981. Evidence for metal poisoning in acute deaths of large red drum *(Sciaenops ocellata)* . *Bull. Environ. Contam. Toxicol* . 27(5):639-644.

Carpenter, J.H. and D.G. Cargo. 1957. The oxygen requirement and mortality of the blue crab in the Chesapeake Bay. Chesapeake Bay Inst., Johns Hopkins Univ., Tech. Rep. XIII, 22 p.

Carr, W.E.S. and C.A. Adams. 1973. Food habits of juvenile marine fishes occupying seagrass beds in the estuarine zone near Crystal River, Florida. *Trans. Am. Fish. Soc.* 102:511-540.

Carriker, M.R. 1959. The role of physical and biological factors in the culture of *Crassostrea* and *Mercenaria* in a salt-water pond. *Ecol. Monogr.* 29:219-266.

Carriker, M.R. 1951. Ecological observations on the distribution of oyster larvae in New Jersey estuaries. *Ecol. Monogr.* 21(1):19-38.

Carriker, M.R. 1952. Some recent investigations on native bivalve larvae in New Jersey estuaries. *Proc. Natl. Shellfish. Assoc.* (1950): 69-74.

Carriker, M.R. 1961. Interrelation of functional morphology, behavior, and autecology in early stages of the bivalve *Mercenaria mercenaria.* *J. Elisha Mitchell Sci. Soc.* 77:168-241.

Carter, R.M. 1968. On the biology and paleontology of some predators of bivalved Mollusca. *Palaeogeography, Palaeoclimatology, Palaeoecology* 4:29-65.

Castagna, M. and P. Chanley. 1973. Salinity tolerance fo some marine bivalves from inshore and estuarine environments in Virginian waters on the western mid-Atlantic coast. *Malacologia.* 12:47-96.

Chaindusse, J. 1992. Pesonal communication from J. Chaindusse, commercial shrimp fisherman in Tampa Bay to R. Hochberg, March 1992.

Chambers, R. 1981. Seasonal feeding an distribution of *Palaemonetes pugio* and *P. vulgaris* in Great Sippewissett salt marsh. *Biol. Bull.* (Woods Hole) 161:324.

Chanley, P. and J.D. Andrews. 1971. Aids for identification of bivalve larvae of Virginia. *Malacologia* 11:45-119.

Chanley, P.E. 1958. Survival of some juvenile bivalves in water of low salinity, *Proc. Natl. Shellfish. Assoc.* 48:52-65.

Chao L.N. and J.A. Musick. 1977. Life history, feeding habits, and functional morphology of juvenile sciaenid fishes in the York River estuary, Virginia. *Fish. Bull.* 75:657-702.

Chapman, C.R. 1959. Oyster drill *Thais haemastoma* predation in Mississippi Sound. *Proc. Natl. Shellfish. Assoc* . 49:87-97.

Chesney, E.J. and E.D. Houde. 1989. Laboratory studies on the effect of hypoxic waters on the survival of eggs and yolk-sac larvae of the bay anchovy, *Anchoa mitchilli. In:* Population biology of bay anchovy in mid-Chesapeake Bay, 184-191. E.D. Houde et al., eds. Center for Environmental and Estuarine Studies, Chesapeake Biol. Lab. Final Rep. to MD Sea Grant. Ref No. (UMCEES) CBL 89-141.

Chester, A.J. and G.W. Thayer. 1990. Distribution of Spotted Seatrout *(Cynoscion nebulosus)* and Gray Snapper *(Lutjanus griseus)* juveniles in seagrass habitats of western Florida Bay. *Bull. Mar. Sci.* 46(2):345-357.

Chestnut, A.F. 1952. Growth rates and movements of hard clams *Venus mercenaria. Proc. Gulf Caribb. Fish. Inst.* 4:49-59.

Chestnut, D.E. 1983. Feeding habits of juvenile spot *Leiostomus xanthurus* (Lacepede) in North Inlet estuary, South Carolina. M.S. Thesis, Univ. of S.C. Columbia, SC 68 p.

Chittenden, M.E., Jr. 1989. Initiation of trawl surveys for a cooperative research/assessment program in the Chesapeake Bay. Final Rep. to Chesapeake Bay Stock Assessment Committee. Virginia Ins. Mar. Sci., Gloucester Point.

Christmas, J.Y. and W. Langley. 1973. Estuarine invertebrates, Mississippi. *In:* Cooperative Gulf of Mexico Estuarine Inventory and Study, Mississippi, 255-319. J.Y. Christmas, ed. Ocean Springs, MS: Gulf Coast Res. Lab.

Chung, K. 1977. Heat resistance of crustaceans and fishes taken from the intake canal of an estuarine power plant and their predicted survival in the discharge canal. Ph.D. dissertation. Texas A&M University.

Chung, K.S. and K. Strawn. 1982. Predicted survival of the bay anchovy *(Anchoa mitchilli)* in the heated effluent of a power plant on Galveston Bay, Texas. *Env. Biol. Fish* 7(1):57-62.

Clark, P.A. and R.W. MacAuley. 1989. Geography and economy of Tampa Bay and Sarasota Bay. Tampa Bay Regional Planning Council. St. Petersburg, FL. *In:* Tampa and Sarasota Bays: Issues, Resources, Status, and Management. E.D. Estevez, ed., NOAA Estuary of the Month Seminar Series No. **11.**

Clark, S.H. 1971. Factors affecting the distribution of fishes in Whitewater Bay, Everglades National Park, Florida. *Florida Sea Grant Tech. Bull.* 8:1-100.

Clime, R.D. 1976. Setting orientation in the oysters *Ostrea edulis L.* and *Crassostrea virginica* Gmelin. Master's Thesis. University of Maine at Orono. 94pp.

Coe, W.R. 1934. Alternation of sexuality in oysters. *Am. Nat.* 68(716):236-251.

Coen, L.D., K.L. Heck, and L.G. Abele. 1981. Experiments on competition and predation among shrimps of seagrass meadows. *Ecology* 62:1484-1493.

Collins, M.R. 1981. The feeding periodicity of striped mullet, *Mugil cephalus L.,* in two Florida habitats. *J. Fish Biol.* 19:307-315.

Collins, M.R. 1985 . Species Profiles: Life histories and environmental requirements of coastal fishes and invertebrates (South Florida) - striped mullet. *U.S.,Fish and Wildlife Service Biol. Rpt.* 82.

Comp, G.S. 1985. A survey of the distribution and migration of the fishes in Tampa Bay. Mote Marine Laboratory. Sarasota, FL. In: Proceedings Tampa Bay Area Scientific Information Symposium. S.F. Treat, J.L. Simon, R.R. Lewis **III,** R.L. Whitman Jr. eds. Sea Grant Project No. **IR** 182-2.

Conservation Consultants, Inc. 1986. Ecological studies at Beacon Key, Tampa Bay, Florida. 22nd Quarterly Rep. Vol. 6. No. 2 to Tampa Electric Co.

Cook, H.L. and M.A. Murphy. 1969. The culture of larval penaeid shrimp. *Trans. Am. fish. Soc.* 98:751-754.

Cooke, C.W. 1945. Geology of Florida. Bull. *Fla. State Geol. Surv .* 29:1-339.

Copeland, B.J. and H.D. Hoese. 1966. Growth and mortality of the American oyster *Crassostrea virginica* in high salinity shallow bays in central Texas. *Publ. Inst. Mar. Sci. Univ. Texas.* 11:149-158.

Copeland, B.J. and M.V. Truitt. 1966. Fauna of the Aransas Pass Inlet, Texas. **II.** Penaeid shrimp postlarvae. *Tex. J. Sci.* 18:65-74.

Costello, T.J. 1963. Pink shrimp life history. *U.S. Fish. Wildl. Serv. Circ.* 161:35-37.

Costello, T.J. and D.M. Allen. 1960. Notes on the migration and growth of pink shrimp *(Penaeus duorarum). Proc. Gulf Caribb. Fish. Inst.* 12: 5-9.

Costello, T.J. and D.M. Allen. 1962. Survival of stained, tagged, and unmarked shrimp in the presence of predators. Proc. *Gulf Caribb. Fish. Inst .* 14:16-20.

Costello, T.J. and D.M. Allen. 1966. Migrations and geographic distribution of pink shrimp, *Penaeus duorarum* , of the Tortugas and Sanibel grounds, Florida. U.S. Fish Wildl. Serv. Fish. Bull. 65:449-459.

Costello, T.J. and D.M. Allen. 1970. Synopsis of biological data on the pink shrimp *Penaeus duorarum duorarum* Burkenroad, 1939. FAO Fish. Rep. 57: 1499-1537.

Costlow, J.D., Jr. 1963. Regeneration and metamorphosis in larvae of the blue crab, *Callinectes sapidus* Rathbun. *J. Exp. Zoo/.* 152:219-227.

Costlow, J.D., Jr. 1967. The effect of salinity and temperature on survival and **metamorphosis** of **megalops of the** blue crab *Callinectes sapidus. Helgol. Wiss. Meeresunters.* 15:84-97.

Costlow, J.D., Jr. and C.G. Bookhout. 1959. The larval development of *Callinectes sapidus* Rathbun reared in the laboratory. *Biol Bull*. (Woods Hole) 116:373-396.

Couch, J.A. 1978. Diseases, parasites, and toxic responses of commercial penaeid shrimps of the Gulf of Mexico and South Atlantic coasts of North America. U.S. Natl. Mar. Fish. Serv. *Fish.* Bull. 76:1-44.

Cowan, J.H. and R.S.Birdsong. 1985. Seasonal occurrence of larval and juvenile fishes in a Virginia Atlantic coastal estuary with emphasis on drums - (Family Sciaendiae). *Estuaries* 8:48-59.

Crabtree, R.E., E.C. Cyr, R.E. Bishop, L.M. Falkenstein, and J.M. Dean. In Press. Age and growth of tarpon, *Megalops atlanticus* , larvae in the eastern Gulf of Mexico with notes on relative abundance and probably spawning areas.

Crabtree, R.E. 1992. Personal communication from R.E. Crabtree, Florida Department of Natural Resources Florida Marine Research Inst. St. Petersburg, FL to R. Hochberg, January 1992.

Crocker, P.A., C.R. Arnold, J.A. DeBoer, and J.D. Holt. 1981. Preliminary evaluation of survival and growth of juvenile red drum *(Scianops ocellata)* in fresh and salt water. *J World Maricul. Soc.* 12(1):122-134.

Culliton, T.J., M.A. Warren, T.R. Goodspeed, D.G. Remer, C.M. Blackwell, and J.J. McDonough. 1990. 50 years of population change along the nation's coasts: 1960-2010. The second report of a coastal trends series. Strategic Assessment Branch, National Oceanic and Atmospheric Administration, Rockville, MD.

Currin, B.M., J.P. Reed, and J.M. Miller. 1984. Growth, production, food consumption, and mortality of juvenile spot and croaker: a comparison of tidal and nontidal nursery areas. *Estuaries* 7(4A):451-459.

Cyr, E.C. 1991. Aspects of the life history of the tarpon, *Megalops atlanticus* , from south Florida. Doctoral Thesis, Univ. of South Carolina.

Dahlberg, M.D. 1972. An ecological study of Georgia coastal fishes. *U.S. Natl. Mar. Fish. Serv. Fish. Bull.* 70:323-353.

Dalton, R. and W. Menzel. 1983. Seasonal gonadal development of young laboratory-spawned southern *(Mercenaria campechiensis)* and northern *(Mercenariamercenaria)* quahogs and their reciprocal hybrids in northwest Florida, U.S.A. *J. Shellfish Res.* 3:11-18.

Darcy, G.H. 1980. Comparison of ecological and life history information on Gobiid fishes, with emphasis on the southeastern United States. NOAA Technical Memorandum NMFS-SEFC-15. 53pp.

Darnell, R.M. 1958. Food habits of fishes and larger invertebrates of Lake Pontchartrain, Louisiana, an estuarine community. *Publ. Inst. Mar. Sci.* Univ. of Texas. 5:353-416.

Darnell, R.M. 1959. Studies of the life history of the blue crab *(Callinectes sapidus* Rathbun) in Louisiana waters. *Trans. Am. Fish. Soc* **.** 88:294-304.

Darnell, R.M. 1961. Trophic spectrum of an estuarine community, based on studies of Lake Pontchartrain, Louisiana. *Ecology* 42(3):553-568.

Davis, H.C. 1953. On food and feeding of larvae of the American oyster, C. *virginica.* Biol. Bull. (Woods Hole) 104(3):334-350.

Davis, H.C. 1958. Survival and growth of clam and oyster larvae at different salinities. Biol. Bull. (Woods Hole) 114(3):296-307.

Davis, H.C. 1960. Effects of turbidity-producing materials in sea water on eggs and larvae of the clam *(Venus (Mercenaria) mercenaria). Biol. Bull.* (Woods Hole) 118:48-54.

Davis, H.C. and A. Calabrese. 1964. Combined effects of temperature and salinity on development of eggs and growth of larvae of *M mercenaria* and *C. virginica.* U.S. Fish Wildl. Serv. Fish. *Bull.* 63(3):643-655.

Davis, H.C. and P.E. Chanley. 1955. Spawning and egg produciton of oysters and clams. Biol. Bull. (Woods Hole) 110:117-128.

Davis, H.C. and P.E. Chanley. 1956. Effects of some dissolved substances on bivalve larvae. *Proc. Natl. Shellfish. Assoc.* 46:59-74.

Dawson, C.E., Jr. 1953. A survey of the Tampa Bay area. *Fla. State Board Conserv. Mar. Lab. Tech. Ser* **.** 8. 39pp.

Dawson, C.E. 1958. A study of the biology and life history of the spot, *Leiostomus xanthurus* Lacepede, with special reference to South Carolina. Bear Bluff Lab. Contrib. 28.

Day, J.W., W.G. Smith, P.R. Wagner, and W.C. Stone. 1973. Community structure and carbon budget of a marsh and shallow bay estuarine system in Louisiana. Louisiana State University Publication LSU-SG-72-04. 80 p.

deFur, P.L., C.P. Mangurn, and J.E. Reese. 1990. Respiratory responses of the blue crab *Callinectes sapidus* to long-term hypoxia. Biol Bull. 1 78:46-54.

deSylva, D.P. 1954. The live bait shrimp fishery of the northeast coast of Florida. Fla. State Board Conserv. *Mar. Res. Lab. Tech. Ser* **.** No. 11. 35 p.

desylva, D.P., F.A. Kalber, Jr., and C.N. Shuster. 1962. Fishes and ecological conditions in the shore zone of the Delaware River Estuary, with notes on other species collected in deeper water. *Univ. Del Mar. Lab. Info. Ser. Publ. No. 5* . 164 pp .

Detwyler, R. and E.D. Houde. 1970. Food Selection by Laboratory-reared Larvae of the Scaled Sardine *Harengula pensacolae* (Pisces, Clupiede) and the Bay Anchovy *Anchoa mitchilli* (Pisces:Engraulidae). *Marine Biology* 7:214-222.

DiDomenico, D.A. and R.L. Iverson. 1977. Uptake of glycolic acid by a marine bivalve. *J. Exp. Mar. Biol. Ecol.* 28:243-254.

Dillon, R.T. and J.J. Manzi. 1988. The genetics and shell morphology in a hybrid zone between the hard clam *Mercenaria mercenaria* and *Mercenaria campechiensis. Mar. Biol.* 100:217-222.

Dobkin, S. 1963. The larval development of *Palaemonetes paludosus* (Gibbes, 1850) (Decapoda, Palaemonidae), reared in the laboratory. *Crustaceana* 6:41-61.

Dobkin. S. 1961. Early developmental stages of pink shrimp, *Penaeus duorarum* , from Florida waters. U.S. Fish Wildl. Serv. Fish. *Bull.* 61:321-349.

Dobkin, S. and R.B. Manning. 1964. Osmoregulation in two species of *Palaemonetes* (Crustacea: Decapoda) from Florida. Bull. *Mar. Sci. Gulf Caribb* . 14:149-157.

Doering, P.H. 1981. Observations on the behavior of *Asterias forbesi* feeding on *Mercenaria mercenaria Ophelia* 20:169-177.

Douderoff, P. 1957. Water quality requirements of fishes and effects of toxic substances. pp. 403-430. *In:* M.E. Brown, Ed. The Physiology of Fishes. Academic Press, New York.

Drew, R.D. 1990. Hydrology and Water Quality, Ch. 4. *In:* Wolfe, S.H. and R.D. Drew (eds.). An Ecological Characterization of the Tampa Bay Watershed. *U.S. Fish and Wildl. Serv. Biol. Rep.* 90(20).

Durako, M.J., M.D. Murphy, K.D. Haddad. 1988. Assessment Fisheries Habitat: Northeast Florida. Florida Marine Research Pub. No. 45.

Edwards, R.E. 1 990. Identification, classification, and inventory of critical nursery habitats for commercially and recreationally important fishes in the Manatee River estuary system of Tampa Bay. Draft Final Project Report. Submitted to SWFWMD, Swim Program, Brooksville, FL.

Eissnberg, M. and J.J. Topping. 1984. Organochlorine residues in shellfish from Maryland Waters 1976-1980. *J Environ. Sci. Health.* B19:673-688.

Eldred 1968. First record of larval tarpon, *Megalops atlanticus* Valenciennes, from the gulf of Mexico. *FL Bd. Conserv. Mar. Lab. Serv.* Vol. IV - Immature Vertebrates.

Eldred, B. 1967. Larval tarpon, *Megalops atlanticus* Valenciennes,(Megalopidae) in Florida waters. *Fl. Bd. Conserv. Mar, Lab. Leafl. Ser.* 4(4) 9-27.

Eldred, B., R.M. Ingle, K.D. Woodburn, R.F. Hutton, and H. Jones. 1961. Biological observations on the commercial shrimp *Penaeus duorarum* Burkenroad, in Florida waters. *Fla. Board Conserv. Mar. Lab. Prof. Pap .* Ser. 3. 139p.

Eldred, B. and W.G. Lyons. 1966. Larval ladyfish, *Elops saurus* Linnaeus 1766, (Elopidae) in Florida and adjacent waters. *Fl. Bd. Conser. Mar. Lab. Leafl. Ser.* 4(2). 6 p.

Eldred, B., J. Williams, G.T. Martin, and E.A. Joyce, Jr. 1965. Seasonal distribution of penaeid larvae and postlarvae of the Tampa Bay area, Florida. *Fla. Board Conserv. Mar. Lab. Tech. Ser.* 44. 47p.

Eldred, B. 1968. First Record of a larval tarpon, *Megalops atlanticus* Valenciennes, from the Gulf of Mexico. *Fla. Bd. Conserv., Mar. Lab. Leafl. Ser.* Vol. IV - Immature Vertebrates.

Ellis, M.J. and B.C. Coull. 1989. Fish predation on meiobenthds: field experiments with juvenile spot *Leiostomus xanthurus* Lacepede. *J. Exp. Mar. Biol. Ecol.* 130:19-32.

Ellis, R.W. 1956. Tarpon Cooperative Research Program - Progress Report. The Marine Laboratory, Univ. of Miami.

Etzold, D.J. and J.Y. Christmas. 1979. A Mississippi Shellfish Management Plan. MS-AL Sea Grant Consortium. MASGP-78-046.

Eversole, A.G. 1986. Gametogenesis and spawning in North American clam populations: implications for culture. *In:* Clam mariculture in North America. J.J. Manzi and M. Castagna, eds. Amsterdam: Elsevier Sci. Publ. Co.

Eversole, A.G. 1987. Species Profiles: Life histories and environmental requirements of coastal fishes and invertebrates (South Atlantic) - hard clam. U.S. Fish Wildl. Serv. Biol. Rep. 82(11.75) U.S. Army Corps of Engineers, TR EL-82-4. 33pp.

Eversole, A.G., W.K. Michener, and P.J. Eldridge. 1980. Reproductive cycle of *Mercenaria mercenaria* in a South Carolina estuary. Proc. Natl. Shellfish. Assoc. 70:22-30.

Ewald, J.J. 1965. The laboratory rearing of pink shrimp, *Penaeus duorarum* Burkenroad. *Bull. Mar. Sci.* 15:436-449.

Farrell, D. 1992. Personal communication from D. Farrell, Florida Department of Environmental Regulation, Tampa, FL to R. Hochberg, March 1992.

Feller, R.J., B.C. Coull, and B.T. Hertschel. 1990. Meiobenthic copepods. Tracers of where juvenile *Leiostomus xanthurus* (Pisces) feed. *Canadian Journal of Fish. and Aq. Sci.* 47:1913-1919.

Finucane, J.H. and R.W. Campbell II. 1968. Ecology of American oysters in Old Tampa Bay, Florida. Q.J. *Fla. Acad. Sci.* 31:37-46.

Fives, J.M., S.M. Warlan, and D.E. Hoss. 1986. Aging and growth of larval bay anchovy, *Anchoa mitchilli*, from the Newport River Estuary, North Carolina. *Estuaries* 9(4B):362-367.

Flannery, M.S. 1989. Tampa and Sarasota Bays' watersheds and tributaries. pp. 18-48. ***In:*** Tampa and Sarasota Bays: Issues, Resources, Status and Management. E.D. Estevez, ed. NOAA Estuary of the Month Seminar Series No. **11.**

Flint, L.H. 1956. Notes on the algal food of shrimp and oysters. Proc. Louisiana Acad. Sci., XIX:11-14.

Florida Department of Natural Resources (FDNR). 1989. Commercial Fisheries Harvest Statistics. Fisheries Statistics Section, FDNR, Florida Marine Research Institute, St. Petersburg, FL.

Florida Fisheries Independent Monitoring Program (FIMP). Unpublished data. Obtained from Bob McMichael, FDNR-FMRI. St. Petersburg, FL.

Florida Fisheries Independent Monitoring Program (FIMP). 1989. 1989 Annual Report. Compiled by Juvenile Fish Group. FL. Mar. Res. Inst. St. Petersburg, FL.

Florida Fisheries Independent Monitoring Program 1990 (FIMP). 1990 Annual Report. Compiled by Florida Dept. of Nat. Res., Florida Mar. Res. Inst., Juvenile fish group, St. Petersburg, FL.

Floyd, W.R. 1977. The effects of temperature and salinity on the larval development of the grass shrimp *Palaemonetes pugio* reared in the laboratory. Va. J. Sci. 28:92.

Fonseca, M. 1992. Unpublished data obtained from M. Fonseca, Natural Marine Fisheries Service, Beauford, NC to K. Killam. February 1992.

Fore, P.L. and T.W. Schmidt. 1973. Biology of juvenile and adult snook, *Centropomus undecimalis,* in the Ten Thousand Islands. pp. SVI-1-SVI-18. *In:* Ecosystems analysis of the Big Cypress Swamp and Estuaries. U.S. Environ. Prot. Agency Surv. Anal. Div., Athens, GA.

Foster, L.M., Jr. 1987. Acute Toxicity Handbook of Chemical to Estuarine Organisms. U.S. EPA, Environ. Res. Lab., Gulf Breeze, FL. EPA/600/8-87/017, 274 p.

Frithsen, J.B. 1989. The benthic communities within Narragansett Bay. An assessment completed for the Narragansett Bay Project. 92p.

Frithsen, J.B., M.C. Fabrizio, J. Gerritsen, A.F. Holland, G.E. Saul, and S.B. Weisberg. 1991b. Example Environmental Assessment Report for Estuaries. EPA/600/05-91/xxx. U.S. Environmental Protection Agency, Atmospheric Research and Exposure Assessment Laboratory, Research Triangle Park, NC.

Frithsen, J.B., K.A. Killam, and M. Young. 1991a. An assessment of key biological resources in the Delaware River Estuary. Submitted to: Delaware Estuary Program; NY, NY. Prepared by Versar, Inc., Columbia, MD.

Funderburk, S.L., S.J. Jordan, J.A. Mihursky, and D. Riley, eds. 1991. Habitat requirements for Chesapeake Bay living resources. 2nd ed. Prepared for Living Res. Subcommittee Ches. Bay Program by Habitat Objectives Workgroup Living Res. Sub. and Ches. Res. Cons. Ins. Solomons, MD.

Fuss, C.M., Jr. and L.H. Ogren. 1966. Factors affecting activity and burrowing habits of the pink shrimp, *Penaeus duorarum* Burkenroad. *Biol. Bull.* (Woods Hole, Mass.) 130:179-191.

Futch, C.R. 1970a. The spotted seatrout, *Cynoscion nebulosus* (Cuvier). Fl. Dept. Nat. Resour. Salt Water Fish. Leafl. 11, 11 p.

Futch, C.R. 1970b. Contributions to the ecology of larval and juvenile lined sole *Achirus lineatus,* in Tampa Bay, Florida. *Mar. Res. Lab. Florida Dept. Nat. Res. Leaflet Series:* Vol. IV - Immature vertebrates, Part 1, No. 17.

Gadbois, D.F. and R.S. Maney. 1983. Survey of polychlorinated biphenyls in selected finfish species from United States coastal waters. *U.S. Natl. Mar. Fish. Serv. Fish. Bull.* 81(2):389-396.

Gallaway, B.J. and K. Strawn. 1974. Seasonal abundance and distribution of Marine fishes at a hot-water discahrge in Galveston Bay, Texas. *Contrib. in Mar. Sci.* 18:71-137.

Gallaway, B.J. and K. Strawn. 1975. Seasonal and areal comparisons of fish diversity indices at a hot-water discharge in Galveston Bay, Texas. *Contrib. Mar. Sci.* 19:81-89.

Galtsoff, P.S. 1961. Physiology of reproduction in molluscs. *Am. Zool.* 1(2):273-289.

Galtsoff, P.S. 1964. The American oyster *Crassostrea virginica Gmelin. U.S. Fish Wildl. Serv. Fish. Bull.* 64. 480 pp.

Gerry, L.R. 1981. The effects of salinity fluctuations and salinity gradients on the distribution of juvenile spot, *Leiostomus xanthurus,* and croaker, *Micropogonias undulatus.* M.S. Thesis. North Carolina State Univ., Raleigh. 57 pp.

Gilmore, R.G. 1987. Subtropical-tropical seagrass communities of southeastern United States: Fishes and fish communities. *Fla. Mar. Res. Publ.* No. 42. Florida.

Gilmore, R.G., L.H. Bullock, and F.H. Berry. 1977. Hypothermal mortality in marine fishes of south-central Florida. *Northeast Gulf Sci.* 2(2):77-97.

Gilmore, R.G., C.J. Donohoe, and D. W. Cooke. **1983.** Observations on the distribution and biology of east-central Florida populations of the common snook, *Centropomus undecimalis* (Bloch). *Fla. Sci. Spec. Suppl*. Issue 45(4) Part 2.

Gilmore, R.G., J.K. Holt, R.S. Jones, G.R. Kulczycki, L.G. Macdowell, **III,** W.C. Magley. 1978. Portable tripod dropnet for estuarine fish studies. *Fish Bull.* 76:285-289.

Ginsburg, R.N. and H.A. Lowenstam. 1958. The influence of marine bottom communities on the depositioned environment of sediments. *J. Geol.* 66(3):310-318.

Godcharles, M.F. 1971. A study of the effects of a commercial hydraulic clam dredge on benthic communities in estuarine areas. *Fla. Dep. Nat. Resour. Mar. Res. Lab., Tech. Ser.* No. 64, 51p.

Godcharles, M.F. and W.C. Jaap. 1973. Exploratory clam survey of Florida nearshore and estuarine waters with commercial hydraulic dredging gear. *Florida Dept. Nat. Res. Profess. Paper Ser.* 21:1-77.

Goodyear, C.P. 1987. Status of the red drum stocks in the Gulf of Mexico. Contrib. CRD 86/87-34, Southeast Fish. Cent. Natl. mar. Fish Serv., NOAA, Miami, FL 33149,49 p.

Goodyear, P.C. 1989. Stock assessment for Gulf of Mexico red drum Project (Grant No. 89NUMFS01) TO MARFIN.

Gosline, W.A. 1971. Functional morphology and classification of teleostean fishes. University Press of Hawaii, Honolulu. 208 p.

Govoni, J.J. and A.J. Chester. 1990. Diet Composition of larval *Leiostomos xanthurus* in and about the Mississippi River plume. *J. Plankton Res.* 12(4):819:830.

Govoni, J.J., A.J. Chester, D.E. Hoss, and P.B. Orther. 1985. An observation of eipsodic feeding and growth of larval *Leiostomus xanthurus* in the northern Gulf of Mexico, *J Plank. Res.* 7:137-146.

Govoni, J.J., D.E. Hoss, and A.J. Chester. 1983. Comparative feeding of three larval fishes in the northern Gulf of Mexico: *Brevoortia patronus, Leiostomus xanthurus,* and *Micropogonias undulatus. Mar. Ecol. Prog. Ser.* 13:189-199.

Gray, E.H. and C.L. Newcombe. 1938. Studies of moulting in *Callinectes sapidus* Rathbun. Growth 2:285-296.

Guillard, R.R. 1957. Some factors in the use of nannoplankton cultures as food for larval and juvenile bivalves. *Proc. Natl. Shellfish. Assoc.* 48: 134-142.

Gunter, G. 1941. Death of fishes due to cold on the Texas coast, January 1940. *Ecology* 22:203-208.

Gunter, G. 1945. Studies on marine fishes of Texas. *Publ. Inst. Mar, Sci. Univ. Tex.* 1:1-190.

Gunter, G. 1950. Distributions and abundance of fishes on the Aransas National Wildlife Refuge, with life history notes. *Pub/. Inst. Mar. Sci.* 1(2):89-101.

Gunter, G. 1953. The relationship of the Bonnet Carre Spillway to oyster beds in Mississippi Sound and the “Louisiana Marsh” with a report on the 1950 opening. *Publ. Inst. Mar. Sci. Univ. Tex.* 3(1):17-71.

Gunter, G. 1955. Mortality of oysters and abundance of certain associates as related to salinity. *Ecology* 36(4):601-605.

Gunter, G. and W.J. Demoran. 1971. Mississippi oyster culture. *Am. Fish Farmer* 2(5):8-12.

Gunter, G., J.Y. Christmas, and R. Killebrew., 1964. Some relations of salinity to population distribution of motile estuarine organisms, with special reference to penaeid shrimp. *Ecology* 45: 181-185.

Gunter, G. and G.E. Hall. 1962. Biological investigations of Caloosahatchee estuary in connection with Lake Okeechobee discharges through Caloosahatchee River. Rep. to dist. Eng. Jacksonville Dist. Corps Eng., 59 p.

Gunter, G. and G.E. Hall. 1963. Biological investigations of the St. Lucie Estuary (Florida) in connection with Lake Okeechobee discharges through the St. Lucie Canal. Gulf Coast *Res. Rep.* 1(5): 189-307.

Gunter, G. and H.H. Hildebrand. 1951. Destruction of fishes and other organisms on the south Texas coast by the cold wave of January 28-February 3, 1951. *Ecology* 32:731-735.

Haddad, K.D. 1989. Habitat trends and fisheries in Tampa and Sarasota Bays. *In:* Tampa and Sarasota Bays-issues, resources, status, and management, 113-128. E.D. Estevez, ed. NOAA estuary of the month. Seminar Series No. 11.

Haddad, K.D., K.A. Killam, B.A. Hoffman, R.H. McMichael, Jr., P. Carlson, G.A. McGarry, R. Hochberg, J.L. Serino. 1987. Assessment of fisheries habitat. Tasks 1, 2, 3, 4, 5. Final Rep for Grant Period 10/86-9/87. Florida Dept. Natl. Res., St. Petersburg, FL.

Haddad, K.D., G.A. McGarry, R.E. Mathewson, Jr., D.A. Rydene, K. M. Peters, M. Durato, F.S. Sargent, P.F. Houhoulas, A.L. Petri, S.D. Giordano, L.A. Houchin, and N.J. Berill. 1990. Marine Resources Geographic Information System and Fishery Resources. Final Report. FL Dept. Nat. Res. FL Mar. Res. Inst., St. Petersburg, FL.

Haddad, K.D., G.A. McGarry, F.J. Sargent, R.E. Matheson, Jr., D.A. Rydene, K.M. Peters, C.A. Friel, H.A. Norris, and T.J. Leary. 1992. Marine Resources Geographic Information System and Fishery Resources. Final Report. Fl. Dept., Nat. Res. Fl. Mar. Res. Inst. St. Petersburg, FL.

Hale, R.C. 1988. Disposition of polycyclic aromatic compounds in blue crabs, *Callinectes sapidus,* from the southern Chesapeake Bay. *Estuaries* 11:255-263.

Hardy, J.D., Jr. 1978. Development of fishes of the Mid-Atlantic Bight: an atlas of egg, larval and juvenile stages. Vol. 2: Anguillidae through Syngnathidae. U.S. Fish Wildl. Serv. Biol. Serv. Program FWS/OBS-78/12. 458 pp.

Harrington, R.W., Jr. and W.L. Bidlingmayer. 1958. Effects of dieldrin on fishes and invertebrates of a salt marsh. *J Wildl. Manag.* 22:76-82.

Harrington, R.W., and E.S. Harrington. 1961. Food selection among fishes invading a high subtropical salt marsh: from onset of flooding through the progress of a mosquito brood. *Ecology* 42:646-666.

Harrington, R.W., Jr. and E.S. Harrington. 1960. Food of larval and young tarpon, *Megalops atlantica. Copeia* 1960:311-319.

Harrington, R.W., Jr. and E.S. Harrington. 1972. Food of female marsh killifish *Fundulus confluentus* in Florida. *Am. Midl. Nat.* 87:492-502.

Harris, B.A., K.D. Haddad, K.A. Steidinger, and J.A. Huff. 1983. Assessment of Fisheries Habitat: Charlotte Harbor and Lake Worth, Florida. Florida Department of Natural Resources, St. Petersburg, FL.

Hartman, D.S. 1979. Ecology and Behavior of the manatee *(Trichechus manatus)* in Florida. Special Publication No. 5. The American Society of Mammalogists.

Haven, D.S., W.J. Hargis, Jr., J.G. Loesch, and J.P. Whitcomb. 1977. The effect of tropical storm Agnes on oysters, hard clams, soft clams, and oyster drills in Virginia. *Chesapeake Res. Consortium Pub .* 54:488-508.

Hayes, P.F. and R.W. Menzel. 1981. The reproductive cycle of early setting *Crassostrea virginica* (Gmelin) in the northern Gulf of Mexico, and its implications for population recruitment. *Biol. Bull.* (Woods Hole) 160:80-88.

Heck, K.L., Jr. and R.J. Orth. 1980. Structural components of eelgrass *Zostera marina* meadows in the lower Chesapeake Bay - Decapod Crustaces. *Estuaries.* 3:289-295. .

Heck, K.L., Jr. and T.A. Thoman. 1981. Experiments on predator-prey interactions in vegetated aquatic habitats. *J. Exp. Mar. Biol. Ecol.* 53:125-134.

Heck, K.L. and T.A. Thoman. 1984. The nursery role of seagrass meadows in the upper and lower reaches of the Chesapeake Bay. *Estuaries* 7:70-92.

Hedgpath, J.W. 1967. Ecological aspects of the Laguna Madre, a hypersaline estuary. pp. 383-389. *In:* G.H. Lauff, ed. *Estuaries.* American Assoc. for the Advancement of Science Publ. No. 83.

Hein, S.H. and J.A. Shepard. 1979. Spawning of spotted seatrout in a Louisiana estuarine ecosystem. Proc. Annu. Conf. Southeast. Assoc. Fish Wildl. Agencies 33:451-465.

Hellier, T.R., Jr. 1962. Fish production and biomass studies in relation to photosynthesis in the Laguna Madre of Texas. Publ. *Inst. Mar. Sci.* Univ. Texas. 8:1-22.

Henderson, J.T. 1929. Lethal temperatures of Lamellibranchiata. *Can. Biol.* 4(25):397-411.

Hesselman, D.M., B.J. Barber, and N.J. Blake. 1989. The reproductive cycle of adult hard clams, *Mercenaria* spp. in the Indian River lagoon, Florida. *J. Shellfish. Res.* 8:43-49.

Hettler, W.F., Jr. 1989. Food habits of juveniles of spotted seatrout and grey snapper in Western Florida Bay. *Bull. Mar. Sci.* 44:155-162.

Hettler, W.F., A.B. Powell, and L.L. Clements. 1978. Laboratory-induced spawning and spot *Leisotomus xanthurus* (Lacepede). In Annual Report of the Southeast Fisheries Center, Beaufort Lab. to U.S. Dept. of Energy, July 1, 1978, p. 351 -356.

Hibbert, C.J. 1976. Biomass and production of a bivalve community on an intertidal mud-flat. *J. Exp. Mar. Biol. Ecol.* 25:249-261.

Hidu, H., W.H. Roosenburg, K.G. Drobeck, A.J. McErlean, and J.A. Mihursky. 1974. Thermal tolerance of oyster larvae *Crassostrea virginica* (Gmelin), is related to power plant operation. *Proc. Natl. Shellfish. Assoc.* 64: 102-1 10.

Hildebrand, H.H. 1955. A study of the fauna of the pink shrimp grounds in the Gulf of Campeche. *Publ. lnst. Mar. Sci. Univ. Tex.* 4: 169-232.

Hildebrand, S.F. 1939. The Panama Canal as a passageway for fishes, with lists and remarks on the fishes and invertebrates observed. *Zoologica* 24:15-45.

Hildebrand, S.F. 1943. A review of the American anchovies (Family Engraulidae). *Bull. Bingham Oceanogr. Collect.* Yale Univ. 8(2): 1-165.

Hildebrand, S.F. 1963a. Family Elopidae. pp. 111-131. *In:* Fishes of the western North Atlantic. Sears Found. Mar. Res. Mem. 1(3).

Hildebrand, S.F. 1963b. Family Engraulidae. p. 152-249. *In:* Fishes of the Western North Atlantic, 630. Y.H. Olsen, ed. Sears Foundation for Marine Research. Bonghan Oceanogr. Lab. Yale Univ.

Hildebrand, S.F. and L.E. Cable. 1930. Development and life history of 14 teleostean fishes at Beaufort, North Carolina. *Bull. U.S. Bur. Fish.* 46:383-488.

Hildebrand, S.F. and W.C. Schroeder. 1928. Fishes of the Chesapeake Bay. *Bull. U.S. Bur. of Fish.* 43:1-366.

Hillsborough County Environmental Protection Commission (HCEPC). 1984. Annual report on environmental quality 1982-1983. Hills. Co., Tampa, FL.

Hines, A.H., T.G. Wolcott, and A.M. Haddon. 1987. Predator-prey interactions of blue crabs in a sub-estuary of central Chesapeake Bay. Smithsonian Environmental Research Center, Portunid Ecology Workshop, Abstract, Edgewater, MD.

Ho, M.-S and P.L. Zubkoff. 1979. The effects of a *Cochlodinium hoterolabatum* bloom on the survival and calcium uptake by larvae of the American oyster, *Crassostrea virginica. ln:* Toxic dinoflagellate blooms. D.L. Taylor and H.H. Seliger, eds. New York: Elsevier.

Hodson, R.G., R.G. Fechelm, and R.J. Monroe. 1981b. Upper temperature tolerance of spot, *Leiostomus xanthurus* , from the Cape Fear River estuary, North Carolina. *Estuaries* 4:345-356.

Hodson, R.G., J.O. Hackman, and C.R. Bennett. 1981a. Food habits of young spots in nursery areas of the Cape Fear River estuary, North Caroina. *Trans. Am. Fish. Soc.* 110:495-501.

Hoese, H.D. 1960. Juvenile penaeid shrimp in the shallow Gulf of Mexico. *Ecology* 41 (3):592-593.

Hoese, H.D. and R.S. Jones. 1963. Seasonality of larger animals in a Texas turtle grass community. *Publ. Inst. Mar. Sci. Univ. Tex.* 9:37-47.

Hoese, H.D. and R.H. Moore. 1977. Fishes of the Gulf of Mexico, Texas, Louisiana and Adjacent Waters. Texas A&M Univ. Press. 327p.

Holland, A.F., N.K. Mountford, M.H. Heigel, K.R. Kaumeyer, J.A. Mihursky. 1980. Influence of predation on infaunal abundance in upper Chesapeake Bay. *Mar. Biol.* 57:221-235.

Holland, A.F., A.T. Shaughnessy, L.C. Scott, V.A. Dickens, J. Gerritsen, and J.A. Ranasinghe. 1989. Long-term benthic monitoring and assessment program for the Maryland portion of Chesapeake Bay: Interpretive Report. Prepared by Versar, Inc., ESM Operations for Maryland Department of Natural Resources, Power Plant Research Program.

Hollis, E.H. 1952. Variations in the feeding habits of the striped bass, *Roccus saxatilis* (Walbsum) in Chesapeake Bay. Bull. Bingham Oceanogr. Collect, Yale Univ. 14:111-131.

Holt, J., R. Godbout, and C.R. Arnold. 1981. Effects of temperature and salinity on egg hatching and larval survival of red drum, *Sciaenops ocellata. U.S. Natl. Mar. Fish. Serv. Fish. Bull.* 79(3):569-573.

Holt, S. and C. Arnold. 1982. Distribution and abundance of eggs, larvae, and juveniles of red drum *(Sciaenops ocellatus)* in seagrass beds in a south Texas estuary. 5th Annu. Larval Fish Conf., La. Coop. Fish. Res. Unit:86.

Holt, S.A., C.L. Kitting, and C.R. Arnold. 1983. Distribution of young red drums among different sea-grass meadows. *Trans. Am. Fish. Soc*. 112:267-271.

Holthuis, L.B. 1952. A general revision of the Palaemonidae (Crustacea: Decapoda, Natantia) of the Americans. II. The subfamily Palaemoninae. Allan Hancock Found. Publ. Occas. Pap. 12:1-396.

Homer, M., P.W. Jones, R. Bradford, J.M. Scoville, D. Morek, N. Kaumeryer, L. Breisch, K. Huddaway, D. Elam, and J.A. Mihursky. 1980. Dermersal fish food habit studies near the Chalk Point Power Plant, Patuxent estuary, Maryland - 1978-1979. Final Rep. to Maryland Power Plant Siting Prog. Univ. of Maryland, Chesapeake Biol. Lab. Ref. No. UMCEES 80-32-CBL.

Homer, M.L. and J.A. Mihursky. 1991. Spot, *Leiostomus xanthurus. In:* Funderburk et al. (eds.). Habitat requirements for Chesapeake Bay Living Reosurces. 2nd edition. pp. 11:1-11: 19. Prepared for Living Resources Subcommittee, Chesapeake Bay Program.

Hopkins, A.E. 1931. Factors influencing the spawning and setting of oysters in Galveston Bay, Tex. *Bull. U.S. Bur. Fish.* 47(3):57-83.

Hopkins, S.H. 1955. Oyster setting on the gulf coast. Proc. *Natl. Shellfish. Assoc.* 45:52-55.

Horton, D.B. and D.W. Bridges. 1973. A Study to Predict the Effects of Thermal Additions in the Bay River and Neuse River Area of North Carolina. OWRT-004-NC, 63 p.

Hoss, D.E., L.C. Coston, and W.F. Hettler, Jr. 1972. Effects of increased temperature on post larval and juvenile estuarine fish. Proceedings 25th Annual Conference Southeastern Association Game and Fish Comm., pp. 635-642.

Hoss, D.E., L. Coston-Clements, D.S. Peters, and P.A. Tester. 1988. Metabolic responses of spot, *Leiostomus xanthurus,* and Atlantic croaker, *Micropogonias undulatus,* larvae to cold temperatures encountered following recruitment to estuaries. *Fish. Bull.* 86:483-488.

Hoss, D.E., W.F. Hettler, Jr., and C. Coston. 1973. Effects of thermal shock on larval estuarine fish - ecological implications with respect to entrainment in power plant cooling systems. *In:* Proceedings of International Symposium on the early life history of fish. J.H.S. Blaxter, ed. Oban, Scotland, pp. 357-371.

Houde, E.D. 1974. Effects of temperature and delayed feeding on growth and survival of larvae of three species of subtropical marine fishes. *Mar. Biol.* 26:271-285.

Houde E.D. and S. Brandt. 1990. Unpublished data from trawling and hydroacoustic surveys. Research supported by NOAA Sea Grant, Univ. of Maryland System, Chesapeake Biological Laboratory, Solomans, MD.

Houde, E.D. and C.E. Zastrow. 1991. Bay Anchovy. *In:* Habitat Requirements for Chesapeake Bay Living Resources, 8-1-13. S.C. Funderburk, et al., eds. Prepared for Living Resources Subcommittee, Chesapeake Bay Program by Habitat Objectives Workgroup, Living Resources Subcommittee and Ches. Res. Cons. Inc. Solornons, MD.

Houde, E.H., C.R. Futch, and R. Detwyler. 1970. Development of the lined sole, *Achirus lineatus,* described from laboratory-reared and Tampa Bay specimens. Dept. of Natl. Res. Tech Ser. No. 62.

Hubschman, J.H. 1975. Larval development of the freshwater shrimp *Palaemonetes kadiakensis* Rathbun under osmotic stress. *Physiol. Zool.* 48:97-140.

Hubschman, J.H. and A.C. Broad. 1974. The larval development of *Palaemonetes intermedius* (Decapoda, Palaemonidae) reared in the laboratory. *Crustaceana* 26:89-103.

Huff, J.A. and S.P. Cobb. 1979. Penaeid and sergestoid shrimps (Crustacea: Decapoda). Fla. Dep. Nat. Resour. Mar. Res. Lab. Mem. Hourglass Cruises 5(4):1-102.

Hughes, D.A. 1969. On the mechanisms underlying tide-associated movements of *Penaeus duorarum. FA0 Fish. Rep.* 57-3:867-874.

Hughes, J.F., J. Sullivan, and R. Shleser. 1972. Enhancement of lobster growth. *Sci.* 177:1110-1111.

Humm, H.J. 1973. Seagrasses, Part IIIC. *In:* A summary of the knowledge of the eastern Gulf of Mexico, **1-10.** J.I. Jones, R.E. Ring, M.O. Rinkel, and R.E. Smith, eds. St. Petersburg: State Univ. Syst. Fla. Inst. Oceanogr.

Hutton, R.R., B. Eldred, K.D. Woodburn, and R.M. Ingle. 1956. The ecology of Boca Ciega Bay with special reference to dredging and filling operations. *Fla. State Bd. Conserv., Tech. Ser.* 17, 87p.

Ingle, R.M., R.F. Hutton, and R.W. Topp. 1962. Results of tagging of salt water fishes in Florida. *Fla. State Bd. Conserv. Mar. Lab. Tech. Ser. No.* 38, 55 p.

Irvine, A.B. 1983. Manatee metabolism and its influence on distribution in Florida. *Biol. Conserv.* 25:315-334.

Irvine, A.B. and H.W. Campbell. 1978. Aerial census of the West Indian Manatee, *Trichechusmanatus,* in the southeastern United States. *J. Mamm.* 59(3):613-617.

Iversen, E.S. and C.P. Idyll. 1960. Aspects of the biology of the Tortugas pink shrimp, *Penaeus duorarum. Trans. Am. Fish. Soc* . 89:1-8.

Iverson, E.S. and A.C. Jones. 1961. Growth and migration of the Tortugas pink shrimp, *Penaeus duorarum* , and changes in the catch per unit of effort of the fishery. *Fla. State Board Conserv. Mar. Res. Lab. Tech. Ser.* No. 34. 30p.

Iverson, E.S. and A.W. Moffett. 1962. Estimation of abundance and mortality of a spotted seatrout population. *Trans. Am. Fish Soc.* 91:395-398.

Iverson, E.S., and D.C. Tabb. 1962. Subpopulations based on growth and tagging studies of spotted seatrout, *Cynoscion nebulosus* in Florida. *Copeia* 1962:544-548.

Jannke, T.E. 1971. Abundance of young sciaenid fishes in Everglades Natural Park, Florida, in relation to season and other variables. Univ. Miami Sea Grant Program. *Sea Grant Tech, Bull.* 11. 128 p.

Jaworski, E. 1972. The blue crab fishery, Barataria estuary. La. State Univ. Cent. Wetl. Resour. Sea Grant Publ. LSU-SG-72-01.

Jaworksi, E. 1982. History and status of Louisiana's soft-shell blue crab fishery. pp. 153-157. *In:* H.M. Perry and W.A. Van Engel (editors), Proceedings Blue Crab Colloquium, Publication 7, Gulf States Mar. Fish. Comm., Ocean Springs, Mississippi.

Johnson, C.W. 1934. List of marine Mollusca of the Atlantic coast from Labrador to Texas. *Proc. Boston Soc. Nat. Hist.* 40(1):204.

Johnson, G.D. 1978. Development of fishes in the Mid-Atlantic Bight. Vol. IV. Carangidae through Ephippidae. *U.S. Fish Wildl. Sew. Bio. Sew. Program* FWS/OBS-78/12. pp. 203-211.

Johnson, W.S., D.M. Allen, M.V. Ogburn, S.E. Stancyk. 1990. Short-term predation responses of adult bay anchovy, *Anchoa mitchilli* to estuarine zooplankton availability. *Mar. Ecol. Prog. Ser.* 64:55-68.

Joint Subcommittee on Aquaculture. 1983. National Aquaculture Development Plan. Vol. 2. Washington, D.C. 196pp.

Jones, A.C, D.E. Dimitriou, J.J. Ewald, and J.H. Tweedy. 1970. Distribution of early developmental stages of pink shrimp, *Penaeus duorarum,* in Florida waters. *Bull. Mar. Sci.* 20:634-661.

Jones, D.S., I.R. Quitmyer, W.S. Arnold, and D.C. Marelli. 1990. Annual shell banding, age, and growth rate of hard clams *(Mercenaria* spp.) from Florida. *J Shellfish. Res.* 9:215-225.

Jones, P.W., F.D. Martin, and J.D. Hardy, Jr. 1978. Development of fishes of the Mid-Atlantic Bight. An atlas of eggs, larvael, and juvenile stages. Acipenseridae through Ictaluridae. *US Fish Wildl. Ser. Biol. Serv. Program.* FWS/OBS-78/12. Vol. 1 :153-163.

Joseph, E.B. 1972. The status of the sciaenid stocks of the middle Atlantic coast. *Chesapeake Sci.* 13:87-100.

Joseph, E.B. and R.W. Yerger. 1956. The fishes of Alligator Harbor, Florida, with notes on their natural history. *Fla. St. Univ. Stud.,* 22:111-156.

Joyce, E.A., Jr. 1965. The commercial shrimps of the northeast coast of Florida. *Fla. State Board Conserv. Mar. Res. Lab. Prof. Pap. Ser .* No. 6.224p.

Keck, R., R. Maurer, and R. Malouf. 1974. Factors influencing the setting behavior of larval hard clams, *Mercenaria mercenaria.* Proc. *Natl. Shellfish. Assoc .* 64:59-67.

Keck, R.T., D. Maurer, and H. Lind. 1975. A.comparative study of the hard clam gonad developmental cycle. *Biol. Bull.* 148:243-258.

Kennedy, F.S., Jr. and D.G. Barber. 1981. Spawning and recruitment of pink shrimp, *Penaeus duorarum,* off eastern Florida. *J Crustacean Biol.* 1:474-485.

Kennedy, V.A., W.H. Roosenburg, M. Castagna, and J.A. Mihursky. 1974. *Mercenaria mercenaria* (Mollusca: Bivalvia): temperature-time relationships for survival of embryos and larvae. *U.S. Natl. Mar. Fish. Serv. Fish. Bull.* 72:l 160-1 166.

Kennedy, V.S. 1991. American oyster, *Crassostrea virginica. In:* Habitat requirements for Chesapeake Bay living resources. Funderburk et al., eds. 2nd ed. Prepared for Living **Res.** Subcommittee Ches. Bay Prog. by Habitat Objectives Workgroup, Living Res. Subcommittee and Chesapeake Res. Can. Ins., Solomons, MD.

Kernehan, R.J., R.E. Smith, M.R. Headrick, and S.L. Mabrey. 1978. Ecological studies in the vicinity of the proposed Summit Power Station, January through December 1977. Ichthyoplankton. Vol. II. Ichthyological Associates, Inc., Summit, Delaware. 688 p.

Kerswill, C.J. 1941. Some environmental factors limiting growth and distribution of the quahaug *Venus mercenaria* L. Ph.D. thesis, University of Toronto.

Kerswill, C.J. 1949. Effects of water circulation on the growth of quahaugs and oysters. *J. Fish. Res. Board Can.* 7(9):545-551 .

Kilby, J.D. 1949. A preliminary report on the young striped mullet *Mugil cephalus L.* in two gulf coastal areas of Florida. *Q.J. Fla. Acad. Sci.* 11:7-23.

Kilby, J.D. 1955. The fishes of two Gulf coastal marsh areas of Florida. Tulane Stud. *Zoo.* 2:175-247.

Kindall, W.N., Jr., J.R. Hall, and C.H. Soloman. 1973. Fishes, macroinvertebrates, and hydrological conditions of upland canals in Tampa Bay, Florida. *Fish. Bull.* 71:155-163.

King, **B.D., III.** 1971. Study of migratory patterns of fish and shellfish through a natural pass. *Tex. Parks Wildl. Dep. Tech. Ser.* 9. 54 p.

Kirby, D.F. and R.E. Knowlton. 1976. Salinity tolerance and sodium balance in the prawn *Palaemonetes pugio* Holthuis. *Am. Zool.* 16:240.

Kjelson, M.A., D.S. Peters, G.W. Thayer, and G.N. Johnson. 1975. The general feeding ecology of postlarval fishes in Newport River estuary. *Fish. Bull.* 73:137-144.

Kleypas, J. and J.M. Dean. 1983. Migration and feeding of the predatory fish. *J. Exp. Mar. Biol. Ecol.* 71:199-209.

Klima, E.F. and D.C. Tabb. 1959. **A** contribution to the biology of the spotted weakfish, *Cynoscion nebulosus* (Cuvier), from northwest Florida, with a description of the fishery. *Fl. Bd. Consent. Mar. Res. Lab. Tech. Ser.* 30, 25 p.

Kneib, R.T. 1987. Seasonal abundance, distribution and growth of post larval and juvenile grass shrimp *(P. pugio)* in Georgia, **USA.** Salt marsh. *Mar. Bio.* 96:215-223.

Kneib, R.T. and A.E. Stiven. 1982. Benthic invertebrate responses to size density manipulations of the common mummichog *Fundulus heteroclitus* in an intertidal saltmarsh. *Ecology* 63:1518-1532.

Knight, C.E. 1966. Mark-recapture experiments. *U.S. Fish Wildl. Sent. Circ.* 246:21-23.

Knowlton, R.E. and D.F. Kirby. 1984. Salinity tolerance and sodium balance in the prawn *Palaemonetes pugio* Holthuis, in relation to other *Palaemonetes* spp. *Comp. Biochem. Physiol.* 77A:425-430.

Knowlton, R.E., and A.B. Williams. 1970. The life history of *Palaemonetes vulgaris* (Say) and *P. pugio* Holthuis in coastal North Carolina. *J. Elisha Mitchell Sci. Soc.* 86:185.

Kostecki, P.T. 1984. Habitat Suitability Index Models: Spotted Seatrout. USFWS FWS/OBS-82/10.75.

Kraeuter, J.N. and M. Castagna. 1980. Effects of large predators on the field culture of the hard clam, *Mercenaria mercenaria.* *Fish. Bull.* 78:538-541.

Kraeuter, J.N., M. Castagna, and R. van Dressel. 1981. Egg size and larval survival of *Mercenaria mercenaria* (L.) and *Argopecten irradians* (Lamarck). *J. Exp. Mar. Biol. Ecol.* 56:3-8.

Kremer, P. 1979. Predation by the ctenophore *Mnemiopsis leidyi* in Narragansett Bay, Rhode island. *Estuaries* 2:97-105.

Kunneke, J.T. and T.F. Palik. 1984. Tampa Bay Environmental Atlas. *U.S. Fish Wild. Serv. Biol. Rep.* 85(15)74 pp and 38 separate maps.

Kutkuhn, J.H. 1966. Dynamics of a penaeid shrimp population and management implications. *U.S. Fish Wildl. Serv. Fish. Bull.* 65:313-338.

Ladd, H.S. 1951. Brackish-water and marien assemblages of the Texas coast, with special reference to mollusks. *Publ. Inst. Mar. Sci. Univ. Tex.* 2(1):125-164.

Landers, W.S. 1954. Notes on the predation of the hard clam *Venus mercenaria* by the mud crab *Neopanope texana. Ecology* 35:422.

Lassuy, D.R. 1983. Species Profiles: Life Histories and Environmental Requirements (Gulf of Mexico) - Spotted Seatrout. USFWS FWS/OBS-82/11.4.

Laughlin, R.A. 1982. Feeding habits of the blue crab, *Callinectes sapidus* Rathbun, in the Apalachicola estuary, Florida. *Bull. Mar. Sci.* 32:807-822.

Leak, J.C. and E.D. Houde. 1987. Cohort growth and survival of bay anchovy, *Anchoa mitchilli,* larvae in Biscayne Bay, Florida. *Mar. Ecol. Prog. Ser.* 37:109-122.

Lee, C.S. and B. Menu. 1981. Effects of salinity on egg development and hatching in grey mullet, *Mugil cephalus L. J. Fish. Biol.* 19:179-188.

Lee, W.Y., G.J. Holt, and C.R. Arnold. 1984. Growth of red drum larvae in the laboratory. *Trans. Am. Fish. Soc.* 113:243-246.

Lefebvre, L.W. and R.K. Frohlich. 1986. Movements of radio-tagged manatees in southwest Florida, January 1985-March 1986. *U.S. Fish and Wild/. Serv., Unpubl. Interim Rep.*

Leffler, C.W. 1972. Some effects of temperature on the growth and metabolic rate of juvenile blue crabs, *Callinectes sapidus* , in the laboratory. *Mar. Biol. (* Berl.) 14:104-110.

Lewis, R.R., III. 1989. Biology and eutrophication of Tampa Bay. *In:* E.D. Estevez (ed.). Tampa and Sarasota Bays. Issues, Resources, Status and Management. NOAA Estuary of the Month Seminar Series No. 11.

Lewis, R.R., **III,** J.M. Carlton. and R. Lombardo. 1984. Algal consumption by the Manatee *(Trichechus manatus* Ll.) in Tampa Bay, Florida. *Florida Scientist* 47(3):189-191.

Lewis, R.R., III, M.J. Durako, M.D. Moffler, and R.C. Phillips. 1985. Seagrass Meadows of Tampa Bay - A Review. Mangrove Systems, Inc. Tampa, FL. *In:* Proceedings Tampa Bay Area Scientific Information Symposium. S.F. Treat, J.L. Simon, R.R. Lewis III, R.L. Whitman Jr. eds. Rep. No. 65. FL Sea Grant.

Lewis, R.R., **III** and E.D. Estevez. 1988. The ecology of Tampa Bay, Florida: An estuarine profile. *U.S. Fish. Wild/. Serv. Biol. Rep.* 85(7-18). 132 p.

Lindall, W.N., Jr., W.A. Fable, Jr., and L.A. Collins. 1975. Additional studies of the fishes, macroinvertebrates, and hydrological conditions of the upland canals in Tampa Bay, Florida. Fish. Bull. 73:81-85.

Lindall, W.N., Jr., J.R. Hall, and C.H. Saloman. 1973. Fishes, macroinvertebrates and hydrological conditions in upland canals in Tampa Bay, *FL. Fish. Bull.* 71:155-163.

Lipcius, R.N. and A.H. Hines. 1986. Variable functional responses of a marine predator in dissimilar homogeneous microhabitats. Ecology 67:1362-1371.

Lipcius, R.N.. E.J. Olmi **III,** and J. van Montrans. 1990. Planktonic availability, molt stage and settlement of blue crab postlarvae. *Mar. Ecol. Prog. Ser.* 58:235-242.

Livingston, R.J. 1982. Trophic organization of fishes in a coastal seagrass system. *Mar. Ecol. Prog. Ser.* 7:1-12.

Livingston, R.J., G.J. Kobylinski, F.G. Lewis, **III,** and P.F. Sheridan. 1976. Long-term fluctuations of epibenthic fish and invertebrate populations in the Apalachicola Bay, *Florida. Fish. BulL* 74(2):311-321.

Lombardo, **R.** and R.R. Lewis, III,1985. A review of commercial fisheries data. pp. 614-634 in J.F. Treat et al. Proceedings Tampa Bay Area Scientific Information Symposium. Rep. No. 65. Florida Sea Grant College.

Long, E.R., D. MacDonald, and C. Cairncross. 1991. Status and trends in toxicants and the potential for their biological effects in Tampa Bay, Florida. NOAA Technical Memorandum. NOS OMA 58.

Loosanoff, V.L. 1936. Sexual phases in the quahog. *Science* 83:287-288.

Loosanoff, V.L. 1937. Seasonal gonadal changes of adult clams, *Venus mercenaria* (L.). *Biol. Bull.* 72(3):406-416 .

Loosanoff, V.L. 1937a. Development of the primary gonad and sexual phases in *Venus mercenaria.* Linnaeus. *Biol. Bull.* 72:389-405.

Loosanoff, V.L. 1953. Behavior of oysters in water of low salinity. *Proc. Natl. Shellfish. Assoc.* (1952)135-151.

Loosanoff, V.L. and H.C. Davis. 1950. Conditioning of *V. mercenaria* for spawning in winter and breeding its larvae in the laboratory. *Biol. Bull.* 98:60-65.

Loosanoff, V.L. and H.C. Davis. 1963. Rearing of bivalve mollusks. *Adv. Mar. Biol.* 1:1-136.

Loosanoff, V.L., H.C. Davis, and P.E. Chanley. 1966. Dimensions and shapes of larvae of some bivalve mollusks. *Malacologia* 4:351-435.

Loosanoff, V.L. and C.A. Nomejko. 1946. Feeding of oysters in relation to tidal stages and to periods of light and darkness. Bio. Bull. (Woods Hole) 90(3):244-264.

Loosanoff, V.L. and C.A. Nomejko. 1951. Existence of physiologically different races of oysters. *Crassostrea virginica* *Biol. Bull.* 101:151-156.

Lorio, W.J. and W.S. Perret. 1980. Biology and ecology of the spotted seatrout, *Cynoscion nebulosus.* pp. 7-13. *In:* R.O. Williams, J.K. Weaver, and F.A. Kalber, eds. Proceedings: Colloquium on the biology and management of red drum and seatrout. *Gulf States Mar. Fish Comm.* No. 5.

Losee, E. 1979. Relationship between larval and spat growth rates in the oyster *Crassostrea virginica.* *Aquaculture* 16(2):123-126.

Lowe, J.E. 1965. Chronic exposure of blue crabs, *Callinectes sapidus* to sublethal concentrations of DOT. Ecology 46:899-900.

Lowery, T.A. and L.G. Tate. 1986. Effect of hypoxia on haemolymph lactate and behavior of the blue crab *Callinectes sapidus* Rathbun in the laboratory and field. *Comp. Biochem. Physiol.* 85A:689-692.

Lux, F.E. and J.V. Mahoney. 1969. First record of the channel bass, *Sciaenops ocellata* (Linnaeus), in the Gulf of Maine. *Copeia* 1969:632-633.

Lyczkowski-Shultz, J., D.L. Ruple, S.L. Richardson, and J.H. Cowan, Jr. 1990. Distribution of fish larvae relative to time and tide in a Gulf of Mexico barrier island pass. Bull. of Mar. **Sci.** 46(3):563-577.

MacInnes, J.R. and A. Calabrese. 1979. Combined effects of salinity, temperature, and copper on embryos and early larvae of the American oyster, *Crassostrea virginica.* *Arch. Environ. Contam. Toxicol.* 8:353-562.

MacKenzie, C.L., Jr. 1977. Predation on hard clam *(Mercenaria mercenaria)* populations. *Trans. Am. Fish. Soc.* 106:530-537.

MacKenzie, C.L., Jr. 1981. Biotic potential and environmental resistance in the American oyster *Crassostrea virginica* in Long Island Sound. *Aquaculture* 22(3):229-268.

Magalhaes, H. 1948. An ecological study of the snails of the genus *Busycon* at Beaufort, North Carolina. *Ecol. Monogr.* 18:377-409.

Maguire, E.J. 1961. The influence of habitat NaCl concentrations on the distribution of two species of *Palaemonetes. La. Acad. Sci.* 24:71-75.

Mahmoudi, B. 1992 Personal communication from B. Mahmoudi, Florida Department of Natural Resources, Florida Marine Research Institute, St. Petersburg, FL to R. Hochberg, January 1992.

Mahood, R.K. 1975. Spotted seatrout in coastal waters of Georgia. *Proc. Ann. Conf. Southeast - Assoc. Game Fish. Comm* 29:195-207.

Major, P.F. 1978. Aspects of estuarine intertidal ecology of juvenile striped mullet, *Mugil cephalus,* in Hawaii. *U.S. Natl. Mar. Fish. Serv. Fish. Bull.* 76:299-313.

Mangum, C.P., S.L. Santos, and W.R. Rhodes, Jr. 1968. Distribution and feeding in the onuphid polychaete *Diopatra cuprea* (Bosc.) *Mar. Biol.* 2:33-40.

Manooch, C.S. 1972. Food habits of adult and yearling striped bass, *Morone saxatilis* (Walbaum) from Albemarle Sound, North Carolina. M.S. Thesis. North Carolina State Univ., Raleigh. 94 p.

Mansueti, R.J. 1960. Restriction of very young red drum *Sciaenops ocellata,* to shallow estuarine waters of Chesapeake Bay during late autumn. *Chesapeake Sci.* 1(3-4):207-210.

Manzi, J.J., M.Y. Bobo, and V.G. Burrell. 1985. Gametogenesis in a population of the hard clam, *Mercenaria mercenaria* (L.), in North Santee Bay, South Carolina. *Veliger* 28(2)186-194.

Marcus, J.M. and T.D. Mathews. 1987. Polychlorinated biphenyls in blue crabs from South Carolina. *Bull. Environ. Contam. Toxicol.* 39:857-862.

Markle, D.F. 1976. The seasonality of availability and movements of fishes in the channel of the York River. Virginia. *Chesapeake Sci.* 17:50-55.

Marshall, A.R. 1958. A survey of the snook fishery of Florida, with studies of the biology of the principal species, *Centropomus undecimalis* (Bloch). *Fla. Bd. Conserv. Mar. Res. Lab. Tech. Ser.* No. 11. 37 p.

Marshall, N. 1954. Factors controlling the distribution of oysters in a neutral estuary. *Ecology* 35(30):322-327.

Martin, R.A. and J.H. Finucane. 1968. Reproduction and ecology of the longnose killifish. *Q.J Fla. Acad. Sci* 31(2):101-111.

Martosubroto, P. 1974. Fecundity of pink shrimp, *Penaeus duorarum* Burkenroad. *Bull. Mar. Sci.* 24:606-627.

Mather, F.J., **III,** 1952. Sport fishes of the vicinity of the Gulf of Honduras, certain Caribbean Islands, and Carmen, Mexico. *Proc. Gulf. Caribb. Fish. Inst.* 4:118-129.

Matheson, E. 1992. Personal communication from E. Matheson, Florida Department of Natural Resources, Florida Marine Research Institute, St. Petersburg, FL to K. Killam, January 1992.

Mayer, F.L., Jr. 1987. Acute toxicity handbook of chemicals to estuarine organisms. U.S. EPA.

McKenney, C.L., Jr. 1979. Ecophysiological studies on the ontogeny of euryplasticity in the cardian shrimp, *Palaemonetes pugio* (Holthuis) and modifications by zinc. Ph.D. diss., Texas A&M Univ.

McKenney, C.L., and J.M. Neff. 1979. Individual effects and interactions of salinity, temperature and zinc on larval development of the grass shrimp *Palaemonetes pugio.* 1. Survival and developmental duration through metamorphosis. *Mar. Biol.* 52:177-188.

McKeown, B.A. 1984. Fish Migration. Timber Press, Beaverton, Ore. 224p.

McLane, W.M. 1955. The fishes of the St. Johns River system. Ph.D. dissertation. Univ. Fl. 361p.

McMichael, R. 1992. Personal communication from R. McMichael, Florida Department of Natural Resources, Florida Marine Research Institute, St. Petersburg, FL to K. Killam, January 1992.

McMichael, R.H., Jr., and K.M. Peters. 1989. Early life history of spotted seatrout *Cynoscion nebulosus* (Pisces:Scianenidae), in Tampa Bay, Florida. *Estuaries* 12(2):98-110.

McMichael, R.H., Jr., K.M. Peters, and G.R. Parsons. 1989. Early life history of the snook, *Centropomus undecimalis,* in Tampa Bay, Florida. *NE* Gulf Sci. 10:113-125.

Menzel, R.W. 1951. Early sexual development and growth of the American oyster in Louisiana waters. *Science* 113(2947):719-721.

Menzel, R.W. 1961. Seasonal growth of the northern quahog. *Mercenaria mercenaria* and the southern quahog, *M. campechiensis,* in Alligator Harbor, Florida. *Proc. Natl. Shellfish. Assoc.* 52:37-46.

Menzel, R.W. 1962. Seasonal growth of northern and southern quahogs, *Mercenaria mercenaria* and *M. campechiensis,* and their hybrids in Florida. *Proc. Natl. Shellfish. Assoc.* 53:111-119.

Menzel, R.W., N.C. Hulings, and R.R. Hathaway. 1966. Oyster abundance in Apalachicola Bay, Florida, in relation to biotic associations influenced by salinity and other factors. *Gulf Res. Rep.* 2(2):73-96.

Menzel, R.W. and F.E. Nicky. 1958. Studies of the distribution and feeding habits of some oyster predators in Alligator Harbor, Florida. Bull. *Mar. Sci. Gulf Caribb.* 8(2):125-145.

Mercer, L.P. 1984. A biological and fisheries profile of red drum, *Sciaenops ocellatus.* Spec. Sci. Rep. 41, NC Dept. Nat. Resour. Community Dev. Div. Mar. Fish., Raleigh, 89 p.

Merrill, A.S. and J.W. Ropes. 1967. Distribution of southern quahogs off the middle Atlantic coast. *Commer. Fish. Rev.* 29(4):62-64.

Merringer, J.V. 1975. Food habits of the weakfish, *Cynoscion nebulosus,* in North Carolina waters. *Chesapeake Sci.* 16:74-76.

Miles, D.W. 1950. The life histories of spotted seatrout, *Cynoscion nebulosus,* and the redfish, *Seiaenops ocellatus. Tex. Game Fish. Comm. Mar. Lab. Annu. Rep.,* 1949-1950.

Miles, D.W. 1951. The life histories of the spotted seatrout, *Cynoscion nebulosus,* and the redfish, *Sciaenops ocellatus* ; sexual development. Annu. Rep. (1950-1951). Tex. Game, Fish and Oyster Comm. Mar. Lab 11 p. Tex. Parks Wildl. Dep., Austin.

Miller, J.M., J.P. Read, and L. Pietrafesa. 1984. Patterns, mechanisms and approaches to the study of migrations of estuarine-dependent fish larvae and juveniles. pp. 205-225. *In:* J.D. McCleave, G.P. Arnold, J.J. Johnson, and W.H. Neill, eds. Mechanisms of migration in fishes. Plenum Press, New York. 544 pp.

Millikin, M.R. and A.B. Willliams. 1984. Synopsis of biological data on the blue crab, *Callinectes sapidus* Rathbun. NOAA tech. Rep. NMFS 1.

Mitchell, M.E. 1989. A comparison of larval fish abundance between a vegetated and non-vegetated habitat with comments on ecological significance of notochord flexion in larval fish. Masters Thesis. Univ. of South Florida, Tampa, FL.

Miyazaki, N., T. Kusaka, and M. Nishiwaki. 1973. Food of *Stenella coeruleoalba.* Sci. Rep. Whales Res. Inst. *Tokyo.* 25:265-275.

Moe, M.A. and G.T. Martin. 1965. Fishes taken in monthly trawl samples offshore of Pinellas County, Florida, with new additions to the fish fauna of the Tampa Bay area. *Tulane Studies in Zool.* 12(4):129-151.

Moffett. A.W. 1961. Movements and growth of spotted seatrout, *Cynoscion nebulosus* (Cuvier) in West Florida. *Florida Board Conserv. Mar. Res. Lab. Tech Ser.* 36:1-35.

Moffett, A.W. and J.E. Randall. 1957. The Roger Firestone tarpon investigation. *Univ. Miami Mar. Lab. Progr. Rep.* 57-22. 18 p.

Moody, W.D. 1950. A study of the natural history of the spotted seatrout, *Cynoscion nebulosus,* in the Cedar Key, Florida area. *Q.J Fl. Acad. Sci.* 12:147-171.

Moore, R.H., 1976. Observations of fishes killed by cold at Port Aransas, Texas, 11-12 January 1973. *Southwest Nat.* 20:461-466.

More, W.R. 1969. A contribution to the biology of the blue crab *Callinectes sapidus Rathbun* in Texas, with a description of the fishery. *Tex. Parks Wildl. Dep., Tech.* Ser. 1, 31 p.

Morgan, M.D. 1980. Grazing and predation of the grass shrimp *Palaemonetes pugio. Limnol. Oceanogr.* 25:896-902.

Morrison, G. 1971. Dissolved oxygen requirements for embryonic and larval development of the hard-shelled clam, *Mercenaria mercenaria. J Fish. Res. Baord Can.* 28:379-381.

Moser, M.L. and L.R. Gerry. 1989. Differential effects of salinity changes on two estuarine fishes, *Leiostomus xanthurus and Micropogonias undulatus. Estuaries* 12:35-41.

Mulholland, R. 1984. Habitat suitability index models: pink shrimp. U.S. Fish Wildl. Serv. FWS/OBS-82/10.76.

Mullholland, R. 1984. Habitat suitability index models: hard clam. U.S. Fish Wildl. Serv. FWS/OBS-82/10.77 21p.

Murphy, M. 1992. Florida Department of Natural Resources, Florida Marine Research Institute. St. Petersburg, FL. Pers. comm. to R. Hochberg, FL. 1992.

Murphy, M.D. and R.D. Taylor. 1990. Reproduction, growth, and mortality of red drum *Sciaenops ocellatus* in Florida Waters. *Fish. Bull.* 88(3):531-542.

1

Murphy, M. (unpubl. data). Preliminary analyses of spotted seatrout life history and population dynamics in Florida. Fl. Mar. Res. Inst. St. Petersburg, FL.

Music, J.L., Jr. and J.M. Pafford. 1984. Population dynamics and life history aspects of major marine sportfishes in Georgia's coastal waters. Ga. Dep. Nat. Resour., Coastal Resour. Div., Coastal Fish. Sect., Final Rep. Study VI. Fed. Aid Proj. F-31.

Myers, A. 1972. C: Tube-worm-sediment relationships of *Diopatra cuprea* (Polychaeta: Onuphidae). *Mar. Biol.* 17:350-356.

Nagabushanam, R. 1961. Tolerance of the prawn, *Palaemonetes vulgaris* (Say), to waters of low salinity. *Sci. Cult.* 27:43.

Nash, C.E., C.M. Kuo, and S.C. McConnel. 1974. Operational procedures for rearing larvae of the grey mullet *(Mugil cephalus L.). Aquaculture* 3: 15-24.

Nash, C.E. and J.R. Sylvester. 1974. Thermal tolerance of eggs and larvae of Hawaiian striped mullet, *Mugil cephalus L. Trans. Am. Fish. Soc.*

National Academy of Sciences. 1974. Water quality criteria 1972. National Academy of Sciences/National Academy of Engineering. EPA E Col. Res. Serv. Wash. **DC** Govn't Printing Office.

Naughton, S.P. and C.H. Saloman, 1978. Fishes of the nearshore zone of St. Andrew Bay, Florida, and adjacent coast. *Northeast Gulf. Sci.* 2:43-55.

Neill, W.H. Environmental Requirements of Red Drum. 1990. *In:* Red Drum Aquaculture. Chamberlain, G.W., R.J. Miget, and M.G. Haby (Compilers)Texas A&M, TAMU-56-90-603. Sea Grant College Program.

Nelson, J.S. 1984. Fishes of the world, 2nd ed. John Wiley and Sons, New York. 523 P.

Nelson, T.C. and H.H. Haskin. 1949. On the spawning behaviour of oysters and of *Venus mercenaria* with special reference ot the effects of spermatic hormones. *Anat. Rec.* 105:484-485.

Nelson, W.R. 1969. Studies on the croaker, *Micropogon undulatus* Linnaeus, and the spot, *Leiostomus xanthurus* Lacepede, in Mobile Bay, Alabama, *J. Mar. Sci. Alabama* 1:4-92.

Neumann, D.A., J.M. O'Connor, and J.A. Jerk, Jr. 1981. Oxygen consumption of white perch *(Morone americanus),* striped bass *(M. saxatilis)* and spot *(Leiostomus xanthurus). Comp. Biochem. Physiol.* 69A:467-478.

1

Newcombe, C.L. 1945. The biology and conservation of the blue crab, *Callinectes sapidus* Rathbun. Virginia Fisheries Laboratory, Educational Series 4: 1-39.

Newell, R.I.E. 1988. Ecological changes in Chesapeake Bay: are they the result of over-harvesting the American oyster, *Crassostrea virginica? In:* Understanding the Estuary: Advances in Chesapeake Bay Research, 536-546. CRC Publication 129, Gloucester Point, VA.

Nordlie, F.G., W.A. Szelistowski, and W.C. Nordlie. 1982. Ontogenesis of osmotic regulation in the striped mullet, *Mugil cephalus* L. *J. Fish. Biol.* 20:79-86.

O'Neil, S.P. and M.P. Weinstein. 1987. Feeding habits of spot, *Leiostornus xanthurus* in polyhaline versus meso-oligohaline tidal creeks and shoals. *Fish.* Bull. 85:785-796,

O'Shea, T.J., C.A. Beck, R.K. Bonde, H.I. Kochman, and D.K. Odell. 1985. An analysis of manatee mortality patterns in Florida, 1976-81. *J. Wild/. Manage.* 49:I-11.

O'Shea, T.J., and H.I. Kochman. 1990. Florida manatees: Distribution, geographically referenced data sets, and ecological 'and behavioral aspects of habitat use: *In:* Report of the Workshop on GIS as an Aid to Managing Habitat for West Indian Manatees in Florida and Georgia, 11-22. J.E. Reynolds and K.D. Haddad, eds. FDNR-FMRI No. 49.

O'Shea, T.J., J.F. Moore, and H.I. Kochman. 1984. Contaminant concentrations in manatees in Florida. *J. Wildl. Manage* 481-748.

Odum, H.T. 1953. Factors controlling marine invasion into Florida fresh waters. Bull, *Mar. Sci. Gulf. Caribb.* 3:134-156.

Odum, W.E. 1970. Utilization of the direct grazing and plant detritus food chains by the striped mullet *Mugil cephalus.* pp. 222-240. *In:* J.J. Steele, ed. Marine Food Chains. Oliver and Boyd, Ltd., Edinburgh, Scotland.

Odum, W.E. 1971. Pathways of energy flow in a south Florida estuary. Ph.D. dissertation. ,Univ. of Miami (Sea Grant Tech. Bull. No. 7). 162 p.

Oesterling, M.J. 1976. Reproduction, growth, and migration of blue crabs along Florida's Gulf Coast. Fla. Sea Grant Coll. Rep SUSF-SG-76-003.

Olmi, E.J. III. 1990. Predation on blue crab megalopae: effects of predator species, habitat and prey density. *Bull. Mar. Sci.* 46:248 (Abstract only).

Orr, P.R. 1955. Heat death: I. Time-temperature relationships in marine animals. *Physiol. Zool.* 28(4):290-294.

Orth, R.J. and K.L. Heck, Jr. 1980. Structural components of eelgrass *Zostera marina* meadows in the lower Chesapeake Bay-Fishes. *Estuaries* 3:278-288.

Orth, R.J. and J. van Montfrans. 1987. Utilization of a seagrass meadow and tidal marsh creek by blue crabs *Callinectes sapidus.* I. Seasonal and annual variations in abundance with emphasis on post-settlement juveniles. *Mar. Ecol. Prog. Ser.* 41 :283-294.

Osburn, H.R., G.C. Matlock, and A.W. Green. 1982. Red drum *(Sciaenops ocellatus)* movement in Texas bays. *Contr. in Marine Sci.* 25:85-97.

Overstreet, R.M. 1983. Aspects of the biology of the red drum, *Sciaenops ocellatus,* In Mississippi. Gulf Res. Rep. (Suppl. 1), p. 45-68.

Owen, H.M. 1953. The relationship of high temperature and low rainfall to oyster production in Louisiana. Bull. *Mar. Sci. Gulf Caribb.* 3(1):34-43.

Pacheco, A.L. 1962a. Age and growth of spot in lower Chesapeake Bay, with notes on distribution and abundance of juveniles in the York River System. *Ches. Sci.* 3:18-28.

Pacheco, A.L. 1962b. Movements of spot, *Leiostomus xanthurus* in the lower Chesapeake Bay. *Ches. Sci.* 3:256-257.

Palmer, R.A. 1974. Studies on the blue crab *(Callinectes sapidus)* in Georgia. Ga. Dep. Nat. Resour., Contrib. Ser. 29, 59 p.

Parker, J.C. 1971. The biology of the spot, *Leiostomus xanthurus* Lacepede, and Atlantic croaker, *Micropogon undulatus* (Linnaeus), in two Gulf of Mexico nursery areas. Sea Grant Publ. No. TAMU-SG-71-21D. Texas A&M Univ., College Station.

Parrish, P.R., J.A. Couch, J. Forester, J.M. Patrick, Jr., and G.H. Cook. 1973. Dieldrin: effects on several estuarine organisms, Proc 27th Ann. Conf. Southeastern Assoc. Game and Fish Comm., P. 427-434.

Pearse, A.S. 1936. Estuarine animals at Beaufort, North Carolina. *J. Hisha Mitchell Sci.* Soc. 52:174-222.

Pearson, J.C. 1928. Natural history and conservation of the redfish and other commercial sciaenids on the Texas Coast. *Bull. U.S. Bur. Fish.* 64:178-194.

Pearson, J.C. 1929. Natural history and conservation of the redfish and other commercially important sciaenids on the Texas coast. *U.S. Bur. Fish. Bull.* 44:129-214.

Peebles, E.B. and S.G. Tolley. 1988. Distribution, growth, and mortality of larval spotted seatrout, *Cynoscion nebulosus:* A comparison between two adjacent estuarine areas of southwest Florida. *Bull. Mar. Sci.* 42(3):397-410.

Peebles, E.B., M.S. Flannery, R.E. Matheson, Jr., and J.P. Rast. 1992. Fish nursery utilization of the Little Manatee River Estuary: Relationships to Physicochemical gradients and the distribution of food resources. In: Proceedings, Tampa Bay Area Scientific Symposium 2, 341-368. S.F. Treat and P.A. Clark, eds.

Peebles, E.B. In Prep. Fish-nursery utilization of the Little Manatee River estuary, Florida: The role of freshwater discharge. Final Report submitted to the Southwest Florida Water Management District, Brooksville, FL.

Peebles, E.B. 1992. Personal communication from E.B. Peebles, University of South Florida to K. Killam, November 1992.

Peebles, E.B. 1992. Draft. Florida Inshore Habitat Trophic Analysis Program: Data Report for period 03/15/91 - 10/15/92 (DNR contract C-6967) submitted to Fl. Mar. Res. Inst. by the University of South Florida, Department of Marine Science.

Perez F.I. 1969. Western Atlantic shrimps of the genus *Penaeus. U.S. Fish Wildl. Serv. Fish. Bull.* 67:461-591.

Perez-Pinzon, M.A. and P.L. Lutz. 1991. Activity related cost of osmoregulation in the juvenile snook *(Centropomus undecimalis). Bull. Mar. Sci.* 48:58-66.

Perret, W.S. 1971. Cooperative Gulf of Mexico estuarine inventory and study, Louisiana. Phase IV, Biology. La. Wildl. Fish. Comm. Project 2-22-R:41-105.

Perret, W.S., J.E. Weaver, R.O. Williams, P.L. Johansen, T.D. McIlwain, R.C. Raulerson, and W.M. Tatum. 1980. Fishery profiles of red drum and spotted seatrout. No. 6. Ocean Springs, MS: *Gulf States Mar. Fish. Comm.* 60 pp.

Perry, H.M. 1975. The blue crab fishery in Mississippi. *Gulf Res. Rep.* 5:39-57.

Perry, H.M., and T.D. McIlwain. 1986. Species profiles: Life histories and environmental requirements of coastal fishes, and invertebrates (Gulf of Mexico) -- blue crab. *U.S. Fish and Wildl. Serv. Biol. Rep.* 82(11.55). U.S. Army Corps of Engineers. TR EL-82-4. 21 p.

Perry, H.M. and K.C. Stuck. 1982. The life history of the blue crab in Mississippi with notes on larval distribution. *In:* H.M. Perry and W.A. Van Engel (editors), Proceedings Blue Crab Colloquium, October 18-19, 1979, p. 17-22.

Peters, K. 1992. Personal communication from K. Peters, Fl. Dept. Nat. Res. Fl. Mar. Res. Inst. to K. Killam, September 1992.

Peters, D.S. and M.A. Kjelson. 1975. Consumption and utilization of food by various postlarval and juvenile fishes of North Carolina estuaries. *In:* Estuarine Research, Vol. I. Chemistry, geology, and the estuarine system, 448-472. El. Cronin, ed. Acad. Press, NY.

Peters, K.M., N.J. Berill, S.D. Giordano, and E.J. Walton. 1992. Assessment of snook recruitment and nursery habitat in Tampa Bay. Abstract, Florida Chapt. of Am. Fish. Soc. Brooksville, FL.

Peters, K.M. and R.H. McMichael, Jr. Unpublished manuscript. Early life history notes on silver perch *(Bairdiella chrysoural,* sand seatrout *(Cynoscion arenarius),* spot *(Leiostomus xanthurusl,* and three kingfish *(Menticirrhus americanus, M. littoralis* and *M. saxatilis)* from Tampa Bay, Florida, FDNR. FMRI.

Peters, K.M. and R.H. McMichael, Jr. 1987. Early life history of *Sciaenops ocellatus* (Pisces:Sciaenidae) in Tampa Bay, Florida. *Estuaries* 10(2):92-107.

Peterson, C.H. 1982. Clam predation by whelks *(Busycon* spp.): Experimental tests of the importance of prey size, prey density, and seagrass cover. *Mar. Biol.* 66:159-170.

Peterson, C.H. and N.M. Peterson, 1979. The ecology of intertidal flats of North Carolina: a community profile. U.S. Fish Wildl. Serv. Bio. Serv. Program FWSIOBS-79/39. 73 pp.

Peterson, M.S. and R.G. Gilmore, Jr. 1991. Eco-physiology of juvenile snook, *Centropomus undecimalis* (Bloch): Life history implications. *Bull. Mar. Sci.* 48:46-57.

Phillips, T.D. 1981. Spawning season of the bay anchovy, *Anchoa mitchilli* (Valenciennes) in Tampa Bay, Florida, determined from egg and larval surveys. *Fla. sci.* 44:21.

Phillips, J.M., M.T. Hugh, J.H. Kerby, and D.P. Moran. 1989. Species profiles: Life histories and environmental requirements of coastal fishes and invertebrates (mid-Atlantic)-Spot. U.S. Fish and Wildl. Serv. Biol. Rep. 82(11-98), U.S. Army Corp. of Engineers. TREL-82-4. 13 p.

Phillips, R.C. 1960. Observations on the ecology and distribution of the Florida seagrasses. Fla. St. Bd. Conserv. Mar. Lab. Prof. Papers Ser. No. 2:1-72.

Piyatiratitivorakul, S. 1988. The life history and bioenergetics relations in the grass shrimp *Palaemonetes pugio.* Ph.D. diss., Univ. of S. Carolina.

Pomeroy, L.R., and R.G. Wiegert. 1981. The ecology of a salt marsh. Springer-Verlag, New York. 271 p.

Powell, A.B. and H.R. Gordy. 1980. Egg and larval development of the spot *Leiostomus xanthurus* (Sciaenidae). *U.S. Natl. Mar. Fish. Serv. Fish. Bull.* 78:701-714.

Prager, M., J.M. McConaugha, C. Jones, P.J. Geer. 1990. Fecundity of blue crab, *Callinectes sapidus* in Chesapeake Bay: biological, statistical, and management considerations. *Bull. Mar. Sci.* 46:170-179.

Pratt, D.M. 1953. Abundance and growth of *Venus mercenaria* and *Callocardia morrhuana* in the relationship to the character of bottom sediments. *J.Mar. Res.* 12:60-74.

Pratt, D.M. and D.A. Campbell. 1956. Environmental factors affecting growth *in V. mercenaria. Limnol. Oceanogr.* 1:2-17.

Price, W.W. and R.A. Schlueter. 1985. Fishes of the littoral zone of McKay Bay, Tampa Bay System, Florida. *Fl. Sci.* 48:83-96.

Provancha, M.J. and C.R. Hall. 1991a. Ecology and life history of the clown goby inhabiting the upper Banana River, Cape Canaveral, FL. *Environ. Biol. Fish.* 31(1):41-54.

Provancha, J.A. and C.R. Hall. 1991b. Observations of associations between seagrass beds and manatees in east central Florida. *Fla. Sci,* 54:87-98.

Public Service Electric and Gas Company. 1984. Bay anchovy: A synthesis information on natural history, with reference to occurrence in the Delaware River and involvement with the Salem Nuclear Generating Station. Salem Nuclear Generating Station 316(b) Demonstration, Appendix V. Newark, NJ.

Quick, J.A., Jr. and J.G. Mackin. 1971. Oyster parasitism *by Labyrinthomyxa marina in* Florida. *Fla. Dep. Nat. Resour. Mar. Res. Lab. Prof. Pap. Ser.* 13:1-55.

Reagan, R.E. 1985. Species Profiles: Life Histories and Environmental Requirements of Coastal Fishes and Invertebrates (Gulf of Mexico) - Red Drum. USFWS *Biol. Report* 82(11.36).

Reid, G.K., Jr. 1954. An ecological study of the Gulf of Mexico fishes in the vicinity of Cedar Key, Florida. *Bull. Mar. Sci. Gulf Caribb.* 4(1):1-94.

Reid, G.K. and H.D. Hoese. 1958. Size distribution of fishes in a Texas estuary. *Copeia* 1958(3):225-231.

Relyea. K.G. 1983. *Bull State Mus. Biol. Sci.* 29(1):27.

Reynolds, J.E., III, B.B. Ackerman, I.E. Beeler, B.L. Weigle, P.F. Houhoulis 1992. Assessment and management of manatees *(Trichechus manatus)* in Tampa Bay, pp. 289-309. */n:* Proceedings Tampa Bay Area Scientific Information Symposium 2. S.F. Treat, P.A. Clark, eds. Available from: Text, Tampa, FL.

Reynolds, J.E., Ill and D.K. Odell. 1991. Manatees and Dugongs. Facts on File Books, New York.

Reynolds, W.W. and M.E. Casterlin. 1979. Thermoregulatory behavior of the pink shrimp *Penaeus duorarum* Burkenroad. *Hydrobiologia* 67:179-182.

Rhoads, D.C. and G. Pannella. 1970. The use of molluscan shell growth patterns in ecology and paleoecology. *Lethaia* 3:143-161.

Richkus, W.A., P. Jacobson, A. Ranasinghe, A. Shaughnessy, J. Chaillou, P. Kazyak, C. DeLisle, R. Batiuk, and D. Packer. 1991. Characterization of the current status of key resource species in the basins of the Chesapeake Bay. Draft Report. Prepared by Versar, Inc. for the U.S. EPA Chesapeake Bay Program.

Rickards, W.L. 1968. Ecology and growth of juvenile tarpon, *Megalops atlanticus,* in a Georgia salt marsh. *Bu//. Mar. Sci.* 18(1):220-239.

Rinckey, G.R. and C.H. Saloman. 1964. Effect of reduced water temperature on fishes of Tampa Bay, Florida. Quart. J. Fla. Acad. Sci. 27(1):9-16.

Rivas, L.R. 1986. Systematic review of the perciform fishes of the genus *Centropomus. Copea* 3:579-611.

Rivas, L.R. 1962. The Florida fishes of the genus *Centropomus,* commonly known as snook. *Q. J. Fla. Acad,. Sci.* 25(1):53-64.

Roberts, K.J. and M.E. Thompson. 1982. Economic elements of commercial crabbing in Lake Pontchartrain and Lake Borgne. Louisiana Sea Grant Publ. LSU-TL-82-001. 19 p.

Robinette, H.R. 1983. Species Profiles: Life histories and environmental requirements of coastal fishes and invertebrates (Gulf of Mexico) - bay anchovy and striped anchovy. USFWS/OBS-82/11.14.

Robins, C.R. 1978. The tarpon - unusual biology and man's impact determine its future. *Mar. Recreational Fish.* 2:105-112.

Robins, C.R., R.M. Bailey, C.E. Bond, J.R. Brooks, E.A. Lachner, R.N. Lea, and W.B. Scott. 1980. A list **of** common and scientific names of fishes from the United States and Canada. *Am. Fish. Soc. Spec. Pub. No.* 12, 174p.

Robison, D.E. 1985. Variability in the vertical distribution of ichthyoplankton in Lower Tampa Bay. *In:* Proceedings of Tampa Bay Area Scientific Information Symposium. S.F. Treat, J.L. Simon, R.R. Lewis, and R.L. Whitman, Jr., eds.

Roegner, C. and R. Mann. 1991. Hard clam. *In:* Habitat Requirements for Chesapeake Bay Living Resources. Funderburk et al., eds. 2nd ed.

Roelofs, E.W. 1954. Food studies of young sciaenid fishes, *Micropogon and Leiostomus* from North Carolina. *Copeia.* Vol. 1954. pp. 151-153.

Rosen, D.E. 1973. Suborder Cyprinodontoidei. pp. 229-262. *In:* D.M. Cohen, N.B. Marshall, A.W. Ebeling, D.E. Rosen, T. Iwamoto, P. Sonoda, S.B. McDowell, W.H. Week III, and L.P. Woods. Fishes of the western North Atlantic. Sears Found. Mar. **Res.** Mem. 1, Pt. 6, Yale University.

Ross, J.L., J.S. Pavela, and M.E. Chittenden, Jr. 1983. Seasonal occurrence of black drum, *Pogonias cromis,* and red drum, *Sciaenops ocellatus,* off Texas. *Northeast Gulf Sci.* 6/11:67-70.

Ross, S.W. 1980. *Leiostomus xanthurus* Lacepede spot. p. 759. *In:* Atlas of North American Freshwater Fishes. *N.C. State Mus. Nat. Hist.,* 854pp.

Rothschild. B.J. 1990. FISHMAP. Final Rep. to Maryland Department of Natural Resources, Univ. of Maryland, Ches. Biol. Lab., Solomons **Isl.**

Rozas, L.P. and C.J. Hackney. 1984. Use of oligohaline marshes by fishes and macrofaunal crustaceans in North Carolina. *Estuaries* 7:213-224.

Rutherford, E.S., T.W. Schmidt, and J.T. Tilmant. 1986. The early life history of spotted seatrout , red drum, grey snapper, and snook in Everglades National Park, Florida. South Florida Research Center Report. SFRC-86/07.

Rutherford, E.S., T.W. Schmidt, and J.T. Tilmant. 1989. Early life history of spotted seatrout, *(Cynoscion nebulosus)* and grey snapper *(lutjanus griseus)* in Florida Bay, Everglades National Park, Florida. *Bull Mar. Sci.* 44:49-64.

Rutherford, E.S., E.B. Thue, and D.G. Buker. 1982. Population characteristics, food habits, and spawning activity of spotted seatrout, *Cynoscion nebulosus,* in Everglades National Park. Report T-644. Homestead, Florida. South Florida Res. Center, 48 p.

Rutherford, E.S., J.T. Tilmant, E.B. Thue, and T.W. Schmidt. 1989. Fishery harvest and population dynamics of spotted seatrout, *Cynoscion nebulosus,* in Florida Bay and adjacent waters. *Bull. Mar. Sci.* 44(1):108-125.

Ryer, C.H. 1987. Temporal patterns of feeding by blue crabs *(Callinectes sapidus)* in a tidal-marsh creek and adjacent seagrass meadow in the lower Chesapeake Bay. *Estuaries* 10:136-140.

Saloman, C. 1968. Diel and seasonal occurrence of pink shrimp, *Penaeus duorarum* Burkenroad, in two divergent habitats of Tampa Bay, FL. Contrib. No. 37. Bureau of Commercial Fisheries Biol. Lab St. Petersburg, FL. *USFWS Spec. Sci. Rep. No.* 561.

Saloman, C.H. 1965. Bait shrimp *(Penaeus duorarum)* in Tampa Bay, Florida - biology, fishery economics, and changing habitat. *U.S. Fish Wild/. Serv. Spec. Sci. Rep. Fish.* 520:1-16.

Saloman, C.H. and J.L. Taylor. 1969. Age and growth of large southern quahogs from a Florida estuary. *Proc. Natl. Shel/fish. Assoc.* 59:46-51.

Sandifer, P.A. 1973. Effects of temperature and salinity on larval development of grass shrimp *Palaemonetes vulgaris* (Decapoda, Caridea). *U.S. Natl. Mar. Fish. Serv. fish. Bull.* 71:115-123.

Sandoz, M. and R. Rogers. 1944. The effect of environmental factors on hatching, moulting, and survival of zoea larvae of the blue crab *Callinectes sapidus* Rathbun. *Ecology* 25:216-228.

Santos, S.L. and J.L. Simon. 1980. Marine soft bottom community establishment following annual defaunation: Larval or adult recruitment? *Mar. Ecol. Prog. Ser.* 2:235-241.

Savage, N.B. 1976. Burrowing activity in *Mercenaria mercenaria* (L.) and *Spisula solidissima* (Dillwyn) as a function of temperature and dissolved oxygen. *Mar. Behav. Physiol.* 3:221-234.

Schlesselman, G.W. 1955. The gulf coast oyster industry of the United States. *Geograph. Rev.* 45(4):532-541.

Schneider, D. 1978. Equalization of prey numbers of migrating shore birds. *Nature* 271: 353-354.

Schoner, N.S. and P. Johnson. 1990. Chapter 6-Fauna, 216-279. *In:* Wolf, S.H. and R.D. Drew (eds.). An Ecological Characterization of the Tampa Bay Watershed. *U.S. Fish and Wildl. Serv. Biol. Rep.* 90(20) 335 p.

Schroeder, W.C. 1924. Fisheries of Key West and the clam industry of southern Florida. Rep. U.S. Comm. Fish. for 1923, Append. 12, Doc. 962. 74pp.

Seaman, W., Jr. and M. Collins. 1983. Species profiles: life histories and environmental requirements of coastal fishes and invertebrates (South Florida) – snook. U.S. Fish Wildl. *Serv.* FWS/OBS-82/11.16. U.S. Army Corps of Engineers, TR EL-82-4. 16 p.

Setzler, E.M. 1977. A quantitative study of the movement of larval and juvenile Sciaenidae and Engraulidae into the estuarine nursery grounds of Doboy Sound, Sapelo Island, Georgia. Ph.D. Thesis, Univ. Georgia, 121 p.

Shafland, P.L. and K.J. Foote. 1983. A lower lethal temperature for fingerling snook, *Centropomus undecimalis. NE Gulf Sci.* 6:175-177.

Shafland, P.L. and D.H. Koehl. 1979. Laboratory rearing of the common snook. Proc. Annu., Conf. Southeast., Assoc., Fish Wildl. Agencies. 33:425-431.

Sheridan, P.F. 1979. Trophic resource utilization by three species of sciaenid fishes in a Northwest Florida estuary. *Northeast Gulf Science,* 3(1):1-15.

Shlaifer, A. 1941. Additional social and physiological aspects of respiratory behavior in small tarpon. *Zoologica* 26:55-60.

Sikora, W.B. 1977. The ecology of *Palemonetes pugio* in a southeastern salt marsh ecosystem, with particular emphasis on production and trophic relationships. Ph.D. diss., University South Carolina, Columbia.

Simmons, E.G. 1957. Ecological survey of the upper Laguna Madre of Texas. *Publ. Inst. Mar. Sci., Univ. Tex.* 4:156-200.

Simmons, E.G. and J.P. Breuer. 1962. A study of redfish, *Sciaenops ocellata* Linnaeus and black drum, *Pogonias cromis* Linnaeus. *Publ. Inst. Mar. Sci. Univ. Tex.* 8:184-211.

Simon, J. 1992. Personal communication from J. Simon, Department of Biology, Univ. S. Fl. Tampa to R. Hochberg, February 1992.

Simon, J.L. and D.M. Dauer. 1977. Reestablishment of a benthic community following natural defaunation. *In:* Ecology of Marine Benthos, 139-154. Univ. of S. Carolina Press.

Simon, J.L. and D.M. Dauer. 1972. A quantitative evaluation of red-tide induced mass mortalities of benthic invertebrates in Tampa Bay, Florida. Environ. Lett. **3:229-**234.

Simpson, D.C. 1954. Two small tarpon from Texas. *Copeia* 1954:71-72.

Simpson, D.G. and G. Gunter. 1956. Notes on habitats, systematic characters and life histories of Texas salt water cyprinodonts. *Tulane Stud. Zool.* 4:115-134.

Sims, H.W., Jr. and R.J. Stokes. 1967. A survey of the hard shell clam *(Mercenaria campechiensis)* (Gmelin) population in Tampa Bay, Florida. Fla. Board Conserv. Mar. Lab., Spec. Sci. Rep. No. 17. 8p.

Smith, D.G. 1980. Early larvae of the tarpon, *Megalops atlanticus* Valenciennes (Pisces: Elopidae), with notes on spawning in the Gulf of Mexico and the Yucatan Channel. *Bull. Mar. Sci.* 30:136-141.

Smith, L.D. and B.C. Coull. 1987. Juvenile spot (Pisces) and grass shrimp predation on meiobenthos in muddy and sandy substrata. *J. Exp. Mar. Bio. Ecol.* 105:123-136.

Snell, E. 1984. Commercial harvest records. National Marine Fisheries Service, Miami, FL.

Sogard, S.M., D.E. Hoss, and J.J. Govoni. 1987. Density and depth distribution of larval gulf menhaden, *Brevoortia patronus,* Atlantic croaker, *Micropogonias undulatus* and spot, *Leiostomus xanthurus* in the northern Gulf of Mexico. *Fish. Bull.* 85:601-610.

Sogard, S.M., G.V.N. Powell, and J.G. Holmquist. 1989. Utilization by fishes of shallow, seagrass-covered banks in Florida Bay: Two diel and tidal patterns. *Environmental Biology of Fishes* 24(2):81-92.

Soto, L., A. Garcia, and A.V. Botello. 1981. Study of the penaeid shrimp population in relation to petroleum hydrocarbons in Campeche Bank. *Proc. Gulf Carib. Fish. Inst.* 33:81-100.

Sparks, A.K., J.L. Boswell, and J.G. Mackin. 1958. Studies on the comparative utilization of oxygen by living and dead oysters. Proc. *Natl. Shellfish. Assoc.* 48:92-102.

Springer, V.G. and K.D. Woodburn. 1960. An ecological study of the fishes of the Tampa Bay area. St. Petersburg, Florida. Florida State Board. Conserv. Mar. **Res.** Lab. Prof. Paper Ser. 1:1-104.

Stanley, J.G. and R. DeWitt. 1983. Species profiles: life histories and environmental requirements of coastal fishes and invertebrates (North Atlantic) –hard clam. U.S. Fish Wildl. Serv. FWS/OBS-82/11.18. U.S. Army Corps of Engineers, TR EL-82-4. 19pp.

Steele, P. 1982. Synopsis of the biology of the blue crab *Callinectes sapidus* Rathbun, in Florida. pp. 29-35. *In:* H.M. Perry and W.A. Van Engel (editors, Proceedings Blue Crab Colloquium, Publ. 7, Gulf States Mar. fish. Comm., Ocean Springs, Mississippi.

Steele, P. 1991. Population dynamics and migration of the blue crab, *Callinectes sapidus* Rathbun, in the eastern Gulf of Mexico. Proc. Gulf Caribb. Fish. Inst. 40th Annual, Curaca, p. 241-244.

Steele, P. 1992. Personal communication from P. Steele, Florida Department of Natural Resources, Florida Marine Research Institute to R. Hochberg, January 1992.

Steele, P. and T.M. Bert. Unpublished manuscript. Population ecology of the blue crab, *Callinectes sapidus* Rathbun in a subtropical estuary: Population structure, aspects of reproduction and habitat partitioning. Fla. Mar. **Res.** Inst., St. Petersburg, FL.

Steele, P. and H.M. Perry (eds.). 1990. The blue crab fishery of the Gulf of Mexico United States: a regional management plan. GSMFC Rep. No. 21. Gulf States Mar. Fish. Comm., Ocean Springs, Mississippi.

Stentel, H.B. 1971. Oysters. *In:* Treatise on invertebrate paleontology. Part N, Vol. 3. Mollusca 6. K.C. Moore, ed. Geol. Soc. Am., Boulder, Colo. and University of Kansas, Lawrence.

Stewart, K.W. 1961. Contributions to the biology of the spotted seatrout *(Cynoscion nebulosus)* in Everglades National Park, Florida. MS. Thesis, Univ. of Miami, Coral Gables, Fl. 103 p.

Stickney, R.R. and M.L. Cueno. 1982. Habitat suitability index models: Juvenile spot. U.S. Fish and Wildl. Serv., FWS/OBS-82/10.20.

Stoner, A.W. 1980. Feeding ecology of *Lagodon rhomboides* (Pisces: Sparidae): variation and functional responses. *Fishery Bull.* 78:337-352.

Storey, M. 1937. The relation between normal range and mortality of fishes due to cold at Sanibel Island, Florida. *Ecology* 18: 10-26.

Storey, M. and E.W. Gudger. 1936. Mortality of fishes due to cold at Sanibel Island, Florida, 1886-1936. *Ecology* 17(4):640-648.

Strenth, N.E. 1976. A review **of** the systematics and zoogeography of the freshwater species of *Palaemonetes* Heller of North America (Crustacea: Decapoda). Smithsonian Contri. Zool. 228

Sulkin, S.D. and C.E. Epifanio. 1975. Comparison of rotifers and other diets for rearing early larvae of the blue crab, *Callinectes sapidus* Rathbun. *Estuarine Coastal Mar. Sci.* 3:109-113.

Swingle, H.A. 1971. Biology of Alabama estuarine areas -cooperative Gulf of Mexico estuarine inventory. *Ala. Mar. Res. Bull.* 5:1-123.

Swingle, W., T. Leary, C. Davis, V. Blomo, W. Tatum, M. Murphy, R. Taylor, G. Adkins, T. McIlwain, and G. Mattock. 1984. Fishery profile of red drum. Gulf of Mexico Fish. Manage., Council and Gulf States Mar. Fish. Comm., Tampa, FL, 164 p.

Sykes, J.E. and J.H. Finucane. 1965. Occurrence in Tampa Bay, Florida, of immature species dominant in Gulf of Mexico commercial fisheries. *Fish. Bull.* 65:369-379.

Sykes, J.E. and J.R. Hall. 1970. Comparative distribution of mollusks in dredged and undredged portions of an estuary with a systematic list of species. *US. Fish Wildl. Serv. Fish. Bull.* 68(2):299-306.

Sylvester, J.R. 1975. Critical thermal maxima of three species of Hawaiian estuarine fish: a comparative study. *J. Fish Biol.* 7:257-262.

Sylvester, J.R., C.E. Nash, and C. E. Emberson. 1974. Preliminary study of temperature tolerance in juvenile Hawaiian mullet *(Mugil cephalus). Fish-Cult.* 36:99-100.

Sylvester, J.R., C.E. Nash, and C.R. Emberson. 1975. Salinity and oxygen tolerances of eggs and larvae of Hawaiian striped mullet, *Mugil cephalus L. J. Fish Biol.* 7:621-629.

Tabb, D.C. 1958. Differences in the estuarine ecology of Florida waters and their effect on populations of spotted weakfish, *Cynoscion nebulosus* (Cuvier and Valenciennes). *Trans. 23rd N. Am. Wildl. Nat. Resour. Conf.* 392-401.

Tabb, D.C. 1960. The spotted seatrout fishery of the Indian River area, Florida. *Fl. Bd Conser. Tech. Ser.* 33, 20 p.

Tabb, D.C. 1961. A contribution to the biology of the spotted seatrout, *Cynoscion nebulosus* (Cuvier), of east-central Florida. *Fl. Bd. Conserv. Mar. Res. Lab. Tech. Ser.* 35. 22p.

Tabb, D.C. 1966. The estuary as habitat for spotted seatrout *(Cynoscion nebulosus).* pp. 59-67. *In:* R.S. Smith, Chairman. A symposium on estuarine fishes. *Am. Fish. Soc.* Spec. Publ. No. 3.

Tabb, D.C., D.L. Dubrow, and A.E. Jones. 1962. Studies on the biology of the pink shrimp, *Penaeus duorarum* Burkenroad, in Everglades National Park, Florida. ***Fla.*** *State Board Conserv. Mar. Res. Lab. Tech. Ser.* No. 37. 32p.

Tabb, D.C. and R.B. Manning. 1961. A checklist of the flora and fauna of northern Florida Bay and adjacent brackish waters of the Florida mainland collected during the period July 1957 through September 1960. *Bull. Mar. Sci. Gulf and Caribb.* 11(4):552-649.

Tagatz, M.E. 1968a. Biology of the blue crab *Callinectes sapidus* Rathbun, in the St. Johns River, Florida. *U.S. Fish Wildl. Sew. Fish. Bull.* 67(1):17-33.

Tagatz, M.E. 1968b. Growth of juvenile blue crabs *Callinectes sapidus* Rathbun, in the St, Johns River, Florida. *U.S. Fish Wildl. Serv. Fish. Bull.* 67(2):281-288.

Tagatz, M.E, 1969. Some relations of temperature acclimation and salinity to thermal tolerance of the blue crab, *Callinectes sapidus. Trans. Am. Fish. Soc* 98:713-716.

Tagatz, M.E. 1973. A larval tarpon, *Megalops atlanticus,* from Pensacola, Florida. *Copeia* 1973:140-141.

Tagatz, M.E., P.W. Borthwick, and J. Forester. 1975. Seasonal effects of leached mirex on selected estuarine animals. *Arch. Environ. Contam. Toxicol.* 3:371-383.

Taniquchi, A.K. 1980. Effects of the salinity, temperature, and food abundance upon survival of spotted seatrout eggs and larvae. Proceedings of the Colloquium on the Biology and Management of Red Drum and Seatrout. *Gulf States Mar. Fish. Comm. Rep.* 5:16 (Abstract).

Tatum, W.M. 1979. The blue crab fishery of Alabama. *In:* Symposium on the natural resources of the Mobile Bay estuary, Alabama, 211-220. H.A. Loyacano and J.P. Smith, ed.

Tatum, W.M. 1982. The blue crab fishery in Alabama. pp. 23-28. *In:* H.M. Perry and W.A. Van Engel (editors), Proceedings Blue Crab Colloquium, Publ. 7, Gulf States Mar. Fish. Comm., Ocean Springs, Mississippi.

Taylor, R.G. and H.J. Grier. 1991. Protandric hermaphrodism in the common snook, *Centropomus undecimalis.* Abstract. FL. Chapter of American Fisheries Soc.

Taylor, J.L., J.R. Hall, and C.H. Saloman. 1970. Mollusks and benthic environments in Hillsborough Bay, Florida. *U.S. Fish Wildl. Serv., Fish Bull.* 68(2):191-202.

Taylor, J.L. and C.H. Saloman. 1970. Benthic project. *In:* Report of the Bureau of Commercial Fisheries Biological Laboratory, St. Petersburg Beach, Florida, fiscal year 1969, 3-10. J.E. Sykes, ed. *U.S. Fish Wildl. Serv., Circ.* 342.

Taylor, J.L. and C.H. Saloman. 1968a. Benthic project. ***In:*** Report of the Bureau of Commercial Fisheries Biological Laboratory, St. Petersburg Beach, Florida, fiscal year 1967, 3-5. J.E. Sykes, ed. *U.S. Fish Widl. Serv., Circ.*

Taylor, J.L. and C.H. Saloman. 1969. Benthic project. *In:* Report of the Bureau of Commercial Fisheries Biological Laboratory, St. Petersburg Beach, Florida, fiscal year 1968, 3-10. J.E. Sykes, ed. *U.S. Fish Wildl. Serv., Circ. 313.*

Taylor J.L. and C.H. Saloman. 1986b. Some effects of hydraulic dredging and coastal development in Boca Ciega Bay, Florida. *U.S. Fish Wildl. Serv., Fish. Bull.* 67(2):213-241.

Taylor, R. 1992. Personal communication from R. Taylor, Florida Department of Natural Resources, Florida Marine Research Institute, St. Petersburg, FL to K. Killam, January 1992.

Taylor, R.G., J.A. Whittington, and H.J. Grier. In Prep. Biology of the common snook from the east and west coasts of Florida. FL. Mar. Mar. Res. Inst. June 1992.

Taylor, R.G., J.A. Whitington, and H.J. Grier. 1992. Age, growth and reproduction of the common snook from two locations in Florida. Abstract. Florida Chapt. of Am. Fish. Soc. Brooksville, FL.

Tenore, K.R., and W.M. Dunstan. 1973. Comparison of feeding and biodeposition of three bivalves at different food levels. *Mar. Biol.* 21:190-195.

Thayer, G.W., M.A. Kjelson, and T.J. Price. 1976. Feeding habits of avian populations utilizing the estuarine area near Beaufort. pp. 356-363. *In:* Atlantic Estuarine Fisheries Center annual report to the Energy Research and Development Agency, U.S. Natl. Mar. Fish. Serv., Beaufort, NC.

Thayer, G.W., D.R.Colby, W.F. Hettler, Jr. 1987. Utilization of the red mangrove prop root habitat by fishes in south Florida. *Mar. Ecol. Prog. Ser.* 35:25-38.

Thayer, G.W., H.H. Stuart, W.J. Kenworthy, J.F. Ustach, and A.B. Hall. 1978. Habitat values of salt marshes, mangroves, and seagrasses for aquatic organisms. *In:* Wetland Functions and Values: The state of our understanding, 235-247. P.E. Greeson, J.R. Clark, eds. Am. Water Resources Assoc., Minneapolis, MN.

Theiling, D.L. and H.A. Loyacano. 1976. Age and growth of red drum from a saltwater marsh impoundment in South Carolina. *Trans. Am. Fish. Soc.* 105:41-44.

Thompson, J.M. 1966. The grey mullets. *Oceanog. and Mar. Biol. Ann. Review* 4:301-335.

Thomson, J.M. 1963. Synopsis of biological data on the grey mullet *Mugil cephalus* Linnaeus 1758. *Aust.* C.S.I.R.O. *Div. Fish Oceanogr. Fish. Synop.* 1.66 p.

Thornton, L.L. 1975. Laboratory experiments on the oxygen consumption and resistance to low oxygen levels of certain estuarine fishes. MS thesis, Univ. of Del., Newark, DE.

Thorp, J.H. 1976. Interference competition as a mechanism of coexistence between two sympatric species of the grass shrimp *Palaemonetes* (Decapoda: Palaemonidae. *J. Exp. Mar. Biol. Ecol.* 25:19-35.

Thorp, J.H., and D.E. Hoss. 1975. Effects of salinity and cyclic temperature on survival of two sympatric species of grass shrimp *(Palaemonetes),* and their relationship to natural distributions. *J. Exp. Mar. Biol. Ecol.* 18:19-28.

Thorson, G. 1955. Modern aspects of marine level-bottom animal communities. *J. Mar. Res.* 14:387-397.

Thue, F.B., E.S. Rutherford, D.G. Buker. 1982. Age, growth and mortality of the common snook, *Centropomus undecimalis,* in Everglases National Park, Florida. Report T-683. Homestead, FL, Everglades National Park, S. Florida Res. Center.

Tinsman, J.C. and D.L. Maurer. 1974. Effects of a thermal effluent on the American oyster. *In:* Thermal Ecology, 223-236. J.W. Gibbons and R.R. Sharitz, eds. Proc. Symp. held at Augusta, GA May 3-5, 1973. Natl. Tech. Inf. Serv. Springfield, VA. ISBN 0-87079-014-X.

Tolley, S.G., E.T. Dohner, and E.B. Peebles. 1987. Occurrence of larval snook, *Centropomus undecimalis* (Bloch), in Naples Bay, Florida. *Fla. Sci.* 50:34-38.

Topp, R. 1963. The tagging **of** fishes in Florida, 1952. Fla. Bd. Conserv. Mar. Res. Lab. Prof. Pap. Serv. 5:76 p.

Tucker, J.W., Jr. and R.G. Hodson. 1976. Early and mid-metamorphic larvae of the tarpon, *Megalops atlantica,* from the Cape Fear River estuary, North Carolina, 1973-74. *Chesapeake Sci.* 17:123-125.

Turner, H.J. Jr. 1953. A review of the biology of some commercial molluscs of the east coast of N. America. *In:* 6th Report on investigations of shellfisheries of Massachusetts DNR. Div. Mar. **Res.** Commonw. Mass. Boston.

Turner, R.E. 1981. Plankton productivity and the distribution of fishes on the southeastern U.S. continental shelf. *Sci.* 214:353-354.

U.S. Bureau of Commercial Fisheries. 1961. Gulf of Mexico shrimp catch. Statistical Branch.

U.S. EPA. 1990. Near Coastal Program Plan for 1990: Estuaries. EPA 600/4-90/033. U.S. Environmental Protection Agency, Environmental Research Laboratory, Office of Research and Development, Narragansett, RI.

U.S. Fish and Wildlife Service. 1991. Endangered and Threatened Wildlife and Plants. 50 CFR 17.11 and 17.12.

United States Dept. of Commerce. 1913. Population-1910. Vol II. Bureau of the Census. Washington Government Printing Office.

Ursin, M.J. 1977. A guide to the fishes of the temperate Atlantic coast. E.P. Dutton, NY. 269 pp.

Valiela, I., J.E. Wright, J.M. Teal, and S.B. Volkman. 1977. Growth, production and energy transformations in the saltmarsh killifish, *Fundulus heteroclitus. Mar. Biol.* (Berl.) 40(2):135-144.

van Montfrans, J., C.A. Peery, and R.J. Orth. 1990. Daily, monthly, and annual settlement patterns by *Callinectes sapidus* and *Neopanope sayi* megalopae on artificial collectors deployed in the York River, Virginia: 198501988. *Bull. Mar. Sci.* 46:214-229.

Van Den Avyle, M.J. and D.L. Fowler. 1984. Species Profiles: Life histories and environmental requirements of coastal fishes and invertebrates blue crab (South Atlantic). U.S. Fish and Wildl. Serv., Publ. FWS/OBS-82/11.19, Washington, DC.

Van Engel, W.A. 1958. The blue crab and its fishery in the Chesapeake Bay. Part 1. Reproduction, early development, growth, and migration. Commer. Fish *Rev.* 20(6):6-17.

Van Engel, W.A. 1982. Blue crab mortalities associated with pesticides, herbicides, temperature, salinity, and dissolved oxygen,. pp. 89-92. *In:* H.M. Perry and W.A. Van Engel (editors), Proceedings Blue Crab Colloquium, Publ. 7, Gulf States Mar. Fish. Comm., Ocean Springs, Mississippi.

Van Heukelem, W.F. 1991. Blue crab: *Callinectes sapidus. In:* Habitat requirements for Chesapeake Bay living resources. 2nd ed. Funderburk et al., eds. Prepared by Habitat Objectives Workgroup and Ches. Res. Cons. Inc. for Chesapeake Bay Program.

Van Heukelem, W.F. and S.D. Sulkin. 1990. Effects of low temperature on activity and survival of juvenile blue crabs. *Bull. Mar. Sci.* 46:251. (Abstract only).

Veal, C.D., W.H. Brown, and W.J. Demoran. 1972. Developments in off-bottom oyster culture in Mississippi. Am. Soc. Agric. Eng. Pap. 72-575. 20pp.

Vernberg, F.J. 1972. Animals. *In:* Marine Ecology, 1492-1526. Vol. I, Part 3. O. Kinne, ed. New York: Wiley.

Vetter, R.D. 1982. Seasonal Metabolic Compensation in Symptric Seatrout: Adaptation to the estuary. *Trans. Amer. Fish. Soc.* 111:193-198.

Virnstein, R.W. 1977. The importance of predation by crabs and fishes on benthic infauna in Chesapeake Bay. *Ecology* 58:1199-1217.

Virnstein, R.W. 1972. Effects of heated effluent on density and diversity of benthic infauna at Big Bend, Tampa, Bay, Florida. MS Thesis. Univ. S. Florida, Tampa, FL.

Volpe, A.V. 1959. Aspects of the biology of the common snook, *Centropomus undecimalis* (Bloch) of southwest Florida. Fla. Bd. Conserv. Mar. Lab. Tech. Ser. No. 31. 37 p.

Vouglitois, J.J., I.W. Able, R.J. Kurtt, and K.A. Tighe. 1987. Life history and population dynamics of the bay anchovy in New Jersey. *Trans. Amer. Fish. Soc.* 116(2): 141-153.

Wade, R.A. 1962. The biology of the Tarpon, *Megalops atlanticus,* and the Ox-eye, *Megalops cyprinoides,* with emphasis on larval development. *Bull. Mar. Sci. Gulf and Caribbean* 12(4):545-622.

Wade, R.A. 1969. Ecology of juvenile tarpon and effects of dieldrin on two associated species. U.S. Fish. Wildl. Serv,. Tech. Pap. 41. 85 p.

Wade, R.A. and C.R. Robins. 1973. The tarpon and its sportfishery: a review of our knowledge and outline of the problem. Proc. Int. Game Fish Conf. 15:16-27.

Wakefield, C.A. and R.L. Colura. 1983. Age and growth of red drum in three Texas bay systems. Proc. *Tex. Chapter Am. Fish. Soc.* 5:77-87.

Walker, R.L. and K.R. Tenore. 1984. The distribution and production of the hard clam, *Mercenaria mercenaria,* in Wassau Sound, Georgia. *Estuaries* 7:19-27.

Walsh, W.A., C. Swanson, C.S. Lee, J.E. Banno, and H. Eda. 1989. Oxygen consumption by eggs and larvae of striped mullet, *Mugil cephalus,* in Relation to development, salinity and temperature. *J. Fish Biol.* 35(3):347-358.

Wang, J.C.S. and R.J. Kernehan. 1979. Fishes of the Delaware estuaries: A guide to early life histories. E.A. Communication. Ecological Analysts, Inc., Towson, MD.

Warlen, S.M. and A.J. Chester. 1985. Age, growth, and distribution of larval spot, *Leiostomus xanthurus,* off North Carolina. *Fish. Bull.* 83:587-599.

Weaver, J.E. and L.F. Holloway. 1974. Community structure of fishes and macrocrustaceans in ponds of a Louisiana tidal marsh influenced by weirs. *Contrib. Mar. Sci.* 18:57-69.

Weigle, B.L., J.E. Reynolds III, G.W. Patton, and J.R. Wilcox. 1988. Manatee *(Trichechus manatusl* winter use of warm water discharges in Tampa Bay. Pages 153-164 in K. Mahadevan et al., eds. Proc. Southeastern Workshop on Aquatic Ecological Effects of Power Generation. December 1986. Rept. 124, Mote Marine Laboratory, Sarasota, Fla. 546 pp.

Weinstein, M.P. 1979. Shallow marsh habitats as primary nurseries for fishes and shellfish, Cape Fear River, North Carolina. U.S. Natl. Mar. Fish. Serv. Fish. Bull. 77:339-357.

Weinstein, M.P. and M.P. Wallers. 1981. Growth, survival and production in young-of-year population of *Leiostomus xanthurus,* Lacepede residing in tidal creeks. *Estuaries* 4(3):185-197.

Weinstein, M.P. and R.W. Yerger. 1976. Electrophoretic investigation **of** subpopulations of the spotted seatrout, *Cynoscion nebulosus* (Cuvier), in the Gulf of Mexico and Atlantic coast of Florida. *Comp. Biochem. Physiol.* 548:97-102.

Weintz, M. 1987. Special Series, the Shrimp Connection: Trawling for trouble. *In:* Florida Sportsman, May 1987, 41 -5Op.

Weiss W.R. and T.D. Phillips. 1985. Meroplankton and the Tampa Bay system. pp. 345-358. *In:* Proceedings, Tampa Bay Area Scientific Information Symposium (May 1982). S.F. Treat, J.L. Simon, R.R. Lewis, III, and R.L. Whitman, Jr., eds. Burgess Publishing Co., Inc., Minneapolis, MD.

Wells, H.W. 1957. Abundance of the hard clam, *Mercenaria mercenaria,* in relation to environmental factors. *Ecology* 38:123-128.

Welsh, W.W. and C.B. Breder, Jr. 1923. Contributions to life histories of Sciaenidae of the eastern United States coast. *Bull. U.S. Bur. Fish.* 39:141-201.

Welsh, B.L. 1975. The role of grass shrimp, *Palaemonetes pugio,* in a tidal marsh ecosystem. *Ecology* 56:513-530.

Whetstone, J.M. and A.G. Eversole. 1977. Predation on hard clams, *Mercenaria mercenaria,* by mud crabs, *Panopeus herbstii. Proc. Natl. Shellfish. Assoc.* 68:42-48.

Widdows, J., R.I.E. Newell, and R. Mann. 1989. Effects of hypoxia and anoxia on survival, energy metabolism, and feeding of oyster larvae *(Crassostrea virginica,* Gmelin). Biol. Bull. 177:154-166.

Williams A.B. 1955. A contribution to the life histories of commercial shrimps (Penaeidae) in North Carolina. *Bull. Mar. Sci. Gulf Carib.* 5:116-146.

Williams, A.B. 1958. Substrates as a factor in shrimp distribution. Limnol. Oceanogr. 3:283-290.

Williams, A.B. 1960. The influence of temperature on osmotic regulation in two species of estuarine shrimps *(Penaeus).* Biol. Bull. (Woods Hole, Mass.) 119:560-571.

Williams, A.B. 1965. Marine decapod crustaceans of the Carolinas. *U.S. Fish. Wildl. Serv., Fish. Bull.* 65:1-298.

Williams, A.B. 1984. Shrimp, lobsters, and crabs of the Atlantic coast of the eastern United States, Maine to Florida. Smithsonian Institution Press, Washington, DC.

Williams, A.B. 1974. The swimming crabs of the genus *Callinectes* (Decapoda: Portunidae). *Fish. Bull.. U.S.* 72:685-798

Williams, A.B. and E.E. Deubler, Jr. 1968. A ten-year study of meroplankton in North Carolina estuaries: assessment of environmental factors and sampling success among bothid flounders and penaeid shrimps. Chesapeake Sci. 9:27-41.

Williams, A.B. and R.L. Wigley. 1977. Distribution of northeastern United States based on specimen at Northeast Fisheries Center, Woods Hole, Massachusetts. NOAA Tech. Rep. NWFS Cir 407: 1-44.

Wilson, K.A., K.W. Able, K.L. Heck, Jr. 1990. Habitat use by juvenile blue crabs: a comparison among habitats in southern New Jersey. *Bull. Mar. Sci.* 46:105-114.

Winget, R.R., C.E. Epifanio, T. Runnels, and P. Austin. 1976. Effects of diet and temperature on growth and mortality of blue crab, *Callinectes sapidus,* maintained in a recirculating culture system. *Proc. Natl. Shellfish. Assoc.* 66:29-33.

Wohlschlag, D.E. and J.M. Wakeman. 1978. Salinity stresses, metabolic responses and distribution of the coastal spotted seatrout, *Cynoscion nebulosus. Contrib. in Marine Science* 21:171-185.

Wolfe, S.H. and R.D. Drew, eds. 1990. An ecological characterization of the Tampa Bay watershed. U.S. Fish and Wildl. Svce., Biological Report 90(20).

Wood, C.E. 1967. Physioecology of the grass shrimp *Palaemonetes pugio,* in the Galveston Bay estuarine system. Contrib. Mar. Sci. Univ. Tex. 12:54-79.

Wood, L. and W.J. Hargis, Jr. 1971. Transport of bivalve larvae in a tidal estuary. In: Fourth European Malacology Symposium, 29-44. D.J. Crisp, ed. Cambridge University Press, Cambridge, U.K.

Woodburn, K.D. 1959. Arvida – Bird Key fill. Fla. State. Bd. Conserv., Mar. Lab., Univ. Miami, 59-46, 6p.

Woodburn, K.D. 1961. Survival and growth of laboratory-reared northern clams *(Mercenaria mercenaria)* and hybrids *(M. mercenaria* x *M. campechiensis)* in Florida waters. *Proc. Natl. Shellfish. Assoc.* 52:21-36.

Woodburn, K.D. 1962. Clams and oysters in Charlotte County and vicinity. Florida Bd. Conserv. Mimeo Rpt. FSBCML #62-12, 29pp.

Woodburn, K.D., **B.** Eldred, E. Clark, R.F. Hutton, and R.M. Ingle. 1957. The live bait shrimp industry of the west coast of Florida (Cedar Key to Naples). Fla. St. Bd. Conserv. Tech. Ser. No. 21:1-33.

Yanez-Arancibia, A., F.A. Linares, and J.W. Day, Jr. 1980. Fish community structure and function in Terminos Lagoon, a tropical estuary in the southern Gulf of Mexico. *In:* Estuarine Perspectives, 465-482. V.S. Kennedy, ed. And. Press NY.

Yokel, B. 1966. A contribution to the biology and distribution of the red drum, *Sciaenops ocellata.* M.S. Thesis, Univ. of Miami, 166 p.

Yokel, B. 1980. A contribution to the biology and distribution of the red drum, *Sciaenops ocellata.* (Abstract). *In:* Proc. Red Drum and Seatrout Colloq. Oct 19-20, 1978. p. 5. *Gulf States Mar. Fish. Comm. No.* ***5,*** 118 p.

Zieman, J.C. 1982. The ecology of the seagrasses of South Florida: A community profile. U.S. Fish Wildl. Serv. FWS/OBS-82/85. 158 p.

Zale, A.V. and S.G. Merrifield. 1989. Species Profiles: Life histories and environmental requirements of coastal fishes and invertebrates (South Florida) - Ladyfish and Tarpon. Fish and Wildl. Ser. Biol. Rep. 82(11.104).

Zein-Eldin, Z.P. and G. W. Griffith. 1966. The effect of temperature upon the growth of laboratory-held post-larval *Penaeus aztecus.* Biol. Bull. (Woods Hole) 131:186-196.